ADOPT-A- RIVER PROGRAMME PHASE II: DEVELOPMENT OF AN IMPLEMENTATION PLAN MANUAL FOR VOLUNTEER MONITORING

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EXECUTIVE SUMMARY

The Adopt-a-River programme was initiated to create awareness amongst all South Africans of the need to care for the country's scarce water resources and to facilitate participation in the protection and management of these resources. Involving communities in volunteer monitoring programmes was viewed as one mechanism through which citizens could learn about rivers and reservoirs and become involved in the protection and management of these water bodies in their particular area. One of the objectives of Phase 2 of the Adopt-a-River programme was therefore to provide support for such activities by developing a simple manual that members of the public could use to monitor the state of a water body in their area.

This manual provides a description of basic water monitoring procedures required for water quality monitoring within rivers and reservoirs. The manual provides an introduction to monitoring and covers topics such as planning a monitoring programme in rivers and in reservoirs, security and water safety, basic equipment, and general preparations and sampling procedures. The manual also describes simple monitoring techniques for visually assessing the state of a river or reservoir, procedures for collecting samples, and protocols for monitoring temperature, water clarity, dissolved oxygen, pH, and collecting samples for chemical and bacteriological analysis in a laboratory.

In addition, guidelines for the establishment of a volunteer monitoring programme are also included. These guidelines will assist in developing a monitoring programme that provides useful information that can be used by the Department of Water Affairs (DWA) and by the volunteers themselves to assess the current and potentially changing status of a water resource.

The manual is concluded with a section on reporting pollution incidents in urban and rural situations.

The purpose of this guideline is to create awareness about the water resources (including rivers, streams and reservoirs) located close to volunteers and to get people involved in the monitoring and the protection of the water resources. The data collected through volunteer programmes will help water resource management initiatives, such as those established by DWA, Catchment Management Agencies and Water User Associations, and interest groups to understand, protect and implement interventions.

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LIST OF ACRONYMS

Acronym	Definition
BOD	Biochemical oxygen demand
DO	Dissolved oxygen
DWA	Department of Water Affairs
EPA	Environmental Protection Agency
NTU	Nephelometric turbidity units

GLOSSARY

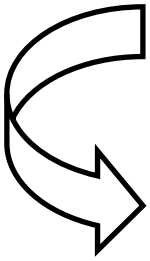
Term	Definition
Algae	Small aquatic plants that occur as single cells, colonies or filaments. They contain chlorophyll but lack specialised water carrying tissues. Through the process of photosynthesis, algae produce most of the food and oxygen in aquatic environments.
Bacteria	A large group of microscopic organisms, generally without chlorophyll. Some are helpful, but certain species are pathogenic and cause diseases such as pneumonia, gastro-enteric illnesses, and typhoid fever, amongst others.
Chlorophyll *a*	A green pigment in algae and other green plants that is essential for the conversion of sunlight, carbon dioxide and water to sugar (see photosynthesis). Sugar is then converted to starch, proteins, fats and other organic molecules. Chlorophyll *a* is a type of chlorophyll present in all types of algae, sometimes in direct proportion to the biomass of algae.
Coliform bacteria	A bacteria carried in human and animal faeces.
Cultural eutrophication	The process of physical, chemical and biological changes associated with enrichment by nutrients, organic matter, silt and sediment associated with human activities that causes a water resource to age unnaturally rapidly. Human activities that result in cultural eutrophication include discharge of sewage, storm water and other nutrient-rich effluents into water bodies from diffuse or point sources.
Decomposition	The transformation of organic molecules (e.g. sugar) to inorganic molecules (e.g. carbon dioxide and water) through biological and non-biological processes.
Diurnal	Refers to an event that occurs in a day (24-hour period) or recurs each day.
Effluent	Liquid wastes from wastewater treatment works, septic systems and/or industrial sources.
Epilimnion	The upper, relatively warm layer of a thermally stratified water column.
Filamentous	Thread-like or hair-like in form.
Flow	The volume of fluid (in this case, water) that passes through a passage of any given section in a unit of time.
Hypolimnion	The lower, cooler layer of a water body during thermal stratification.
Indicator organisms	A wide variety of pathogenic viruses, protozoa and bacteria may be transmitted by water. Infections are generally contracted by drinking contaminated water, recreation exposure to contaminated water, inhaling contaminated aerosols or the consumption or raw food (irrigated vegetables and shellfish) exposed to polluted water. Assessment of the safety of water by tests for the many pathogens which may be present would be impractical for technical and economic reasons. Therefore indicator organisms are used for routine monitoring for potential presence of pathogens in water.
Metalimnion	The middle layer of water that marks the transition between the top (epilimnion) and bottom layers (hypolimnion), where temperature changes rapidly with depth.
Pathogenic	A micro organism capable of producing disease in a host. They are of great concern to human health issues focusing on water bodies.

Term	Definition
Photosynthesis	A chemical reaction that occurs only in plants. Plants use chlorophyll to convert water and carbon dioxide into cellular material and oxygen in the presence of light.
Plankton	Small, mostly microscopic plants (phytoplankton) and animals (zooplankton) that are too small to outswim most currents so the flow of the water tends to move them from place to place.
Reservoir	A water body created by artificially damming a stream or river, where water is collected and kept in quantity for a variety of uses, that may include flood control, water supply, recreation and hydroelectric power.
Riparian	Living or located on the banks of a water body.
Run-off	That portion of precipitation that does not filter into soil but flows over it until ultimately meeting streams, rivers, reservoirs and other water bodies.
Stratification	The process in which several layers of water of different density may form within a water body, usually a reservoir. During stratification the bottom layer is cooler, higher in nutrients and lower in light, productivity and dissolved oxygen. The top layer is warmer, normally lower in nutrients, higher in light, productivity and dissolved oxygen. The boundary between the two is called a thermocline.
Thermal stratification	The stratification of a water body caused by temperature created differences in water density.
Water resource	Includes a watercourse, surface water, estuary or aquifer.
Zooplankton	Microscopic animals that float freely in water, grazing on detritus particles, bacteria and algae, and which may themselves be consumed by fish.

Manual Roadmap

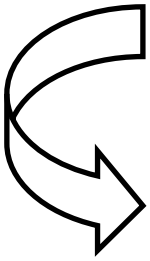
Part 1:
How to use this manual

This chapter provides an overview of the contents of the manual and provides an indication of where to find information.



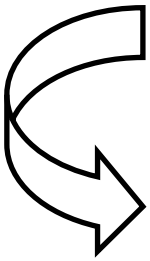
Part 2:
Introduction to Monitoring

This chapter describes how to plan a monitoring programme, it includes security and water safety tips and provides a list of basic sampling equipment.



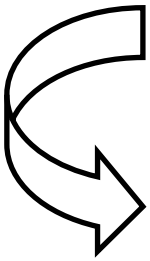
Part 3:
Water quality monitoring

This chapter describes the water quality sampling procedures for both rivers and reservoirs. Information on the different parameters that can be monitored for and the monitoring procedures are also included.



Part 4:
Stream flow conditions inventory

This chapter describes a simple method for measuring stream flow.



Part 5:
Reporting pollution incidents

This chapter provides contact details who should be contacted to report a pollution incident and what information should be provided.

PART 1 : HOW TO USE THE MANUAL

This chapter describes how the manual should be used and where to find appropriate information.

1. PURPOSE OF THE MANUAL

The purpose of this manual is to provide volunteers with sufficient information to develop a monitoring programme which meets their respective data requirements. This manual includes information on how to develop a monitoring programme for both rivers and reservoirs, a description of the types of variables that should be monitored, how best to monitor for these variables and details for who to contact in the event of a pollution incident.

2. HOW TO USE THIS MANUAL

This manual has five sections:

Section 1 outlines how to use the manual and the content included in the manual.

Section 2 provides information on how to plan a monitoring programme. It also includes some basic security and water safety measures that need to be adhered to when going out to monitor in rivers and reservoirs. A list of basic equipment is also provided. A description of general preparation of the sample bottles and sampling procedures is also included.

Section 3 contains information on the water quality monitoring. This section is divided into the monitoring requirements for rivers and reservoirs. Examples of volunteer monitoring programmes are also included in this section.

Section 4 provides a description of stream flow monitoring and estimating the flow of the stream.

Section 5 of this manual contains the contact details for who to contact for further information or in the event of an emergency.

If you want to…….	**Then…..**
Find out how to develop a monitoring programme	Go to Part 2, section 1.1
Find out what basic equipment is required	Go to Part 2, section 3
Find out what variables should be monitored in a river and how to monitor for these variables	Go to Part 3, section 1
Find out what variables should be monitored in a reservoir and how to monitor for these variables	Go to Part 3, section 2
Find out about other monitoring programmes	Go to Part 3, section 3
Find out how to measure stream flow	Go to Part 4, section 1
Find out how to report pollution incidents	Go to Part 5, section 2
Find out about generic water quality limits for various water user categories	Go to Appendix 1
Find out how to make a turbidity tube	Go to Appendix 2
Find out how to make a Secchi disc	Go to Appendix 3

PART 2 : INTRODUCTION TO MONITORING

This chapter provides an introduction to monitoring streams, rivers and reservoirs and what issues need to be considered when designing a volunteer monitoring programme.

1. PLANNING A MONITORING PROGRAMME

Before beginning a monitoring programme, volunteer program officials should structure the programme in such a manner that it produces results that are useful and that answer the requirements of the monitoring programme. The process for designing a monitoring programme is illustrated in **Figure 1.1**. This process is applicable to both volunteer monitoring programmes and formal monitoring programmes.

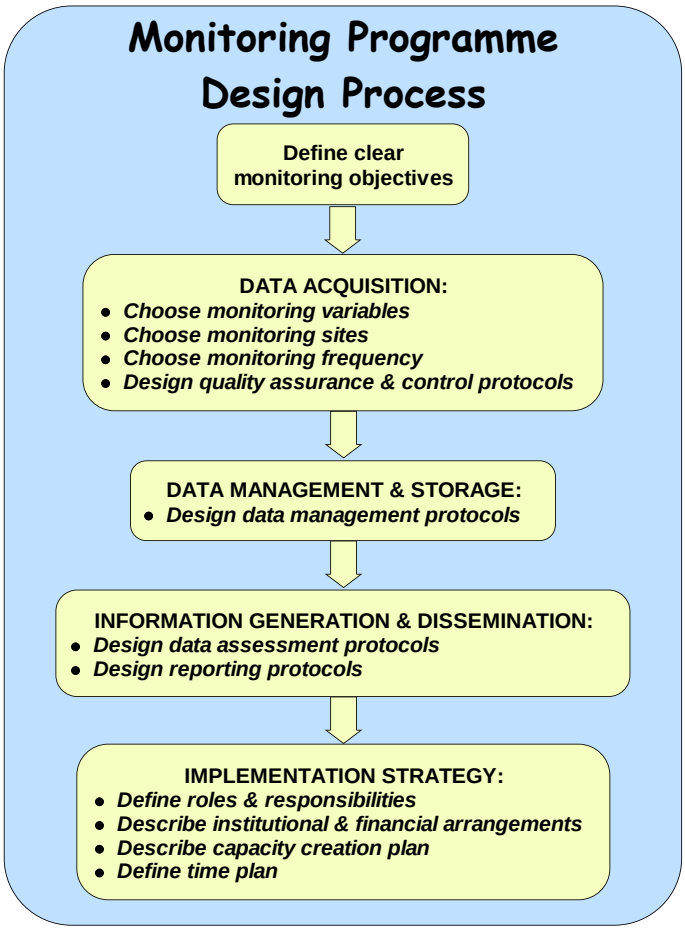


Figure 1.1: The generic process for designing a water quality monitoring programme (from DWAF, 2006).

Define clear monitoring objectives

One of the first steps in planning a monitoring programme is determining why monitoring is taking place. Typical reasons for initiating a volunteer monitoring project include:

- Developing baseline characterisation data;
- Documenting water quality changes over time;
- Screening for potential water quality problems;

- Determining whether waters are safe for swimming;
- Providing a scientific basis for making decisions on the management of a stream or Catchment;
- Determining the impact of a municipal sewage treatment facility, industrial facility or land use activity such as forestry or farming;
- Educating the local community or stream users to encourage pollution prevention and environmental stewardship; and
- Showing public officials that local citizens care about the condition and management of their water resources.

Monitoring strategies

The second step involves selecting a monitoring strategy that is applicable to the type of monitoring that is being undertaken. It is important to understand what the objectives of the monitoring programme are, as this will to a large extent determine which monitoring strategy to adopt. Two of the major strategies are described below:

(1) Ambient (Status) monitoring: The purpose of an ambient monitoring programme is to describe current status or long-term trends in water quality. Many water quality parameters are influenced by the change of seasons or by short-term weather patterns. In order to distinguish between short-term anomalies in the data and actual water quality trends, the parameters need to be measures at consistent intervals over long periods of time.

(2) Baseline monitoring: The purpose of baseline monitoring is to describe baseline conditions in a water resource. Baseline conditions are those which exist before some event that affects water quality occurs, such as a new development or a new discharge from an industry or a wastewater treatment works. Comparing data before and after the event is one way of assessing the impact on water quality.

(3) Compliance monitoring: The purpose of compliance monitoring is to assess whether specific standards or requirements are being met. Compliance monitoring is often used in reference to assess compliance with water use authorisations.

Most volunteer monitoring programmes are designed determine the status of a river, reservoir or wetland.

Monitoring data users

The next step is to identify who will be using the monitoring data. Potential data users include:

- Department of Water Affairs (DWA);
- Local and Regional Municipalities;
- Local and National Environmental Authorities;
- The volunteers themselves;
- Universities;
- Schools; and
- Environmental organisations and/or interest groups.

It is also important to understand how the data will be used. The range of uses of data is unlimited. It could be used to influence local planning to identify a site suitable for a sewage treatment facility, highlight water quality problems and seek solutions or it could be used to educate school children about the importance of water resources.

Monitoring sites

The location of the monitoring sites might be chosen for any number of reasons, including accessibility, proximity to the volunteers' homes or monitoring of potential problem areas. When selecting monitoring sites, the following questions should be asked:

- Are other groups (Local and/or Regional Municipality, DWA, other volunteer groups, schools or universities) already monitoring this site?
- Can you identify the site on a map and on the ground?
- Is the site representative of the Catchment?
- Does the site have water in it during the times of year that monitoring will take place?
- Is there safe, convenient access to the site (including adequate parking) and a way to safely sample a flowing section of the stream? Is there access all year long?
- Do you need permission from the landowner to access the site?
- Can you perform all the monitoring activities and tests that are planned at this site?
- Is the site far enough downstream of drains or tributaries? Is the site near tributary inflows, dams, bridges, or other structures that may affect the results?
- Have you selected enough sites for the study you want to do? The number of monitoring sites will be determined with the type of monitoring strategy that has been adopted (i.e. ambient and baseline monitoring will require several monitoring sites in order to provide information representative of the river or reservoir. While only one or two monitoring points may suffice for compliance monitoring).

Monitoring frequency

The monitoring programme should also specify when monitoring should be conducted and the frequency of the monitoring. The following questions should be considered when determining the frequency of the monitoring events:

- What time of day is best for sampling? For example, temperature and dissolved oxygen can fluctuate naturally as the sun rises and aquatic plants release oxygen.
- What time of year is best for sampling? For example, there is no point in sampling faecal coliform bacteria at swimming beaches in the winter when no one is swimming, or sampling intermittent streams during the dry season, when there is no or low flow in the stream.
- How frequently should monitoring take place? It is possible, for example, to conduct too many biological assessments of a stream and thereby deplete the stream's aquatic community. A program designed to determine whether polluted runoff is a problem would do well to monitor after storms and heavy rainfall.

Data management and storage

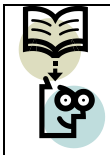
The volunteer program coordinator should have a clear plan for dealing with the data collected each year. Field and lab data sheets should be checked for completeness, data should be screened for outliers, and a database should be developed or adapted to store and manipulate the data. The elements of such a database should be clearly explained in order to allow users to interpret the data accurately and with confidence. Program coordinators will also have to decide how they want to present data results, not only to the general public and to specific data users, but also to the volunteers themselves. Different levels of analysis might be needed for different audiences. A volunteer group collecting data for local or national government use should consult with the appropriate institutions before investing in computerized data management software because the agency could have specific needs or recommendations based on its own data management protocols.

Information generation and dissemination

Raw data should be interpreted in a way that is appropriate for the target audience, it should add value to the raw data, it should be presented in a way that it is understandable by all readers, and ensure that misinterpretation is avoided (DWAF, 2006). Raw data should be compared to water quality guidelines that put it into context. The South African Water Quality Guidelines for Freshwater (2nd edition, 1996) and Coastal Marine Waters (1st edition, 1995) provides threshold levels or ranges associated with certain effects relative to the fitness for use of the water. Although these guidelines do not take account of site-specific situations, they are a useful starting point that gives an approximate indication of the possible effects on water users.

Implementation strategy

An implementation strategy or plan describes who would do the monitoring. It should consider who will undertake the physical monitoring, what monitoring and safety equipment is required, travel arrangements, notification of property owners if samplers need to enter private property, contact lists, etc. The implementation plan deals with the logistical arrangements of executing a monitoring programme.



Additional reading material:

- South African Water Quality Guidelines for Freshwater (2nd edition, 1996)
- South African Water Quality Guidelines for Coastal Marine Waters (1st edition, 1995)

1.1 CONSIDERATIONS FOR MONITORING RIVERS AND STREAMS

Rivers exhibit quite distinct changes in flow, water quality, and health of its ecosystem along it length. These are affected by the landuse in the catchment of the river, the location and size of tributaries flowing into the river, effluent being discharged or seeping into the river, etc. Normally the objective of river or stream monitoring is to describe those changes and understand how the river or stream got into that state. The location and impacts of different types and landuse, tributaries and pollution sources should therefore be considered when designing a monitoring programme and the location of sampling points. One should at least monitor the state of the river where it enters and leaves the area you are interested in. To determine the impacts of tributaries and pollution sources one should locate sampling points downstream of those sources, and if possible, sample the tributaries or pollution sources. Ultimately designing the monitoring network is a compromise between what information you expect to gather and what is possible given the financial and logistical constraints of a volunteer monitoring group. It is better to start with a small monitoring programme and to expand it as you learn more about the behaviour of the river and your group develop better skills and are better resourced.

1.2 CONSIDERATIONS FOR MONITORING RESERVOIRS

Reservoirs exhibit distinct changes in quality and physical characteristics such as water clarity and temperature along its length and with depth. The quality and appearance of reservoirs differ along its length. The upper reaches of the dam is often more like the river flowing into it (for example it's more turbid) whereas the main basin near the dam wall might be clear. Deep reservoirs in South Africa often show distinct differences between the quality of the water near the surface of the dam and near the bottom of the dam. If it is important to understand the changes in quality along the length of the dam, then samples should be collected near the inflow and near the outflow or dam wall, and perhaps some points can be located somewhere between the inflow and the dam wall. If it is important to understand the changes in quality

with depth, then samples should be collected near the surface and the bottom of the dam and measurements such as temperature and oxygen can be taken from the surface to the bottom at short intervals (0.5 to 1 metre).

2. SECURITY AND WATER SAFETY

One of the most critical considerations for a volunteer monitoring program is the safety of its volunteers. All volunteers should be trained in safety procedures and should carry with them a set of safety instructions and the phone number of their program coordinator or team leader. Safety precautions can never be overemphasized. The following are some basic common sense safety rules.

At the site:

- Always monitor with at least one partner. Teams of three or four people are best. Always let someone else know where you are, when you intend to return and what to do if you do not come back at the appointed time.
- Develop a safety plan. Find out the location and telephone number of the nearest police station telephone and write it down. Locate the nearest medical centre and write down directions on how to get between the centre and your site(s) so that you can direct emergency personnel. Have each member of the sampling team complete a medical form that includes emergency contacts, medical aid information and pertinent health information, such as allergies, diabetes, epilepsy, etc.
- Have a first aid kit handy. Know any important medical conditions of team members (e.g. heart conditions or allergic reactions to bee stings). It is best if at least one team member has first aid/CPR training.
- Listen to weather reports. Never go sampling if severe weather is predicted or if a storm occurs while at the site.
- Never wade in swift or high water. Do not monitor if the stream is at flood stage.
- If you drive, park in a safe location. Be sure your car doesn't pose a hazard to other drivers and that you don't block traffic.
- Put your wallet and keys in a safe place, such as a watertight bag you keep in a pouch strapped to your waist.
- Never cross private property without the permission of the landowner. Better yet, sample only at public access points such as bridge or road crossings or public parks.
- Confirm that you are at the proper site location by checking maps, site descriptions, or directions.
- Watch for irate dogs, farm animals, wildlife (particularly snakes) and insects such as ticks, bees and wasps. Know what to do if you get bitten or stung.
- Never drink the water in a stream. Assume it is unsafe to drink and bring your own water from home. After monitoring, wash your hands with antibacterial soap.
- Wear waders and latex gloves in streams suspected of having significant pollution problems.
- Do not monitor if the stream is posted as unsafe for body contact. If the water appears to be severely polluted, contact your program coordinator.
- Do not walk on unstable stream banks. Disturbing these banks can accelerate erosion and might prove dangerous if a bank collapses. Disturb streamside vegetation as little as possible.
- Be very careful when walking in the stream itself. Rocky-bottom streams can be very slippery and can contain deep pools; muddy-bottom streams might also prove treacherous in areas where mud, silt, or sand have accumulated in sink holes. If you must cross the

stream, use a walking stick to steady yourself and to probe for deep water or muck. Your partner(s) should wait on dry land ready to assist you if you fall. Do not attempt to cross streams that are swift and above the knee in depth.

- If you are sampling from a bridge, be wary of passing traffic. Never lean over bridge rails unless you are firmly anchored to the ground or the bridge with good hand/foot holds.
- If at any time you feel uncomfortable about the condition of the stream or your surroundings, stop monitoring and leave the site at once. Your safety is more important than the data!
- If you are sampling in a stream, it is a good idea to make a walking stick of a know length that can be used for balance, prodding and measurement.

Appropriate clothing:

- Be prepared. If it looks like heavy rain, cancel or postpone the monitoring until after the weather improves. If it's cold and it could rain, wear warm clothing, a raincoat and sturdy waterproof shoes. If it's sunny, wear a hat and apply sun screen. If the site has dense vegetation, wear long pants and a long sleeved shirt to avoid scratches.
- When entering shallow waters, make sure you are wearing warders, boots or shoes with a good grip. Do not risk injury by going into the water barefoot, you may cut you feet on sticks or broken glass.
- Bring extra clothes and a towel in case someone slips in and gets wet.

Sampling equipment and procedures:

- Know your equipment, sampling instructions and procedures before going out into the field. Prepare labels and clean equipment before you get started.
- Keep all equipment and chemicals away from small children. Many of the chemicals used in monitoring are poisonous.
- Avoid contact between chemical reagents and skin, eye, nose, and mouth. Never use your fingers to stopper a sample bottle. When you are shaking the sample bottle, always replace the cap before you start shaking the bottle.
- Wear safety goggles when performing any chemical test or handling preservatives.
- Know chemical cleanup and disposal procedures. Wipe up all spills when they occur. Return all unused chemicals to your sampling coordinator for safe disposal.
- Close all containers tightly after use. Do not switch caps.
- Know how to use and store chemicals. Do not expose chemicals or equipment to temperature extremes or long term direct sunshine.
- Do not dispose of used chemicals by dumping them on the ground or in the water resource! Bring a container with a tight fitting lid so that wastes can be returned to a laboratory for proper disposal.

Boat safety equipment checklist:

Before leaving shore, samplers must confirm that all required safety equipment is on board. Boat safety is a subject that samplers need to take seriously because they will be moving around the boat, leaning over the edge and working with equipment.

Samplers should wear a life jacket at all times. Samplers should educate themselves about safe boating rules applicable to the reservoir and must comply with legislative requirements appropriate to the reservoir.

Confirm that the following boat safety equipment is on board the sampling boat:

- Life jacket for each person. Life jackets must be SABS approved, readily available and the proper size;
- First aid kit; and
- Other equipment that may be required by the authority responsible for boating e.g. boats may be required to carry a fire extinguisher and anchor. Also, the boat must be registered according to legislative requirements.

3. BASIC EQUIPMENT

Much of the equipment a volunteer will need is easily obtained from hardware stores or found around the house. Listed below is some basic equipment appropriate for any volunteer field activity. Much of this equipment is optional but will enhance the volunteers' safety and effectiveness.

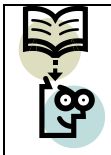
- Laminated map of the study area, clearly indicating the location of the monitoring points. Alternatively a GPS can be used if the monitoring points have been saved as waypoints;
- Boots or waders and life jackets, if you are sampling by boat;
- Walking stick of known length for balance, probing, and measuring;
- Rope / anchor;
- Latex gloves to guard against contamination;
- Insect repellent;
- Sunscreen and a hat;
- Small first aid kit, flashlight, and extra batteries;
- Refreshments, drinking water and anti-bacteria soap;
- Bottle with clean water;
- Clipboard, preferably with plastic cover;
- Several pencils and pens;
- Masking tape to mark sampling bottles;
- Tape measure;
- Thermometer;
- Field data sheet;
- Information sheet with safety instructions, site location information, and numbers to call in emergencies;
- Camera to document particular conditions;
- Cooler box with ice packs;
- Sampling equipment crate; and
- Sampling bottles, which have been cleaned beforehand.

PART 3 : WATER QUALITY MONITORING

This chapter provides background information on various water quality parameters that can be monitored for and it describes how to undertake simple monitoring of water quality in rivers and in reservoirs.

1. WATER QUALITY MONITORING IN RIVERS

Generic water quality limits for the various water user categories (domestic use, irrigation, recreational use and aquatic ecosystems) are provided in **Appendix 1**. These water quality limits provide an understanding of the water quality results in terms of its "fitness for use"[1].



Additional reading material:

- United States Environmental Protection Agency. Volunteer Stream Monitoring: A Methods Manual. http://www.epa.gov/owow/monitoring/volunteer/stream/
- Michaud, J.P. 1991. A Citizen's Guide to Understanding and Monitoring Lands and Streams. http://www.ecy.wa.gov/programs/wq/plants/management/joysmanual/index.html
- Myre, E. and Shaw, R. 2006. The Turbidity Tube: Simple and Accurate Measurement of Turbidity in the Field. www.cee.mtu.edu/sustainable_engineering/resources/technical/Turbidity-Myre_Shaw.pdf
- Waterwatch Australia. Module 2: Getting Started: the team, monitoring and site. http://www.waterwatch.org.au/publications/module2/index.html
- Waterwatch Australia. Module 3: Biological Parameters. http://www.waterwatch.org.au/publications/module3/index.html
- Waterwatch Australia. Module 4: Physical and Chemical Parameters. http://www.waterwatch.org.au/publications/module4/index.html

The following water quality monitoring procedures are discussed in this section:

1. Visual;
2. Temperature;
3. Water clarity / turbidity;
4. Dissolved oxygen;
5. Sampling for chemical analysis;
6. Sampling for bacteriological analysis; and
7. pH.

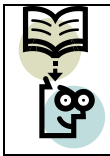
Each of the monitoring procedures are described on a separate page, so that you can add information to a monitoring procedure or you can replace a monitoring procedure as newer information becomes available.

[1] "Fitness for use" of water is a judgment as to how suitable the quality of water is for its intended use.

1.1 VISUAL ASSESSMENT

Visual monitoring provides a qualitative description of the physical characteristics of the river and river banks. Depending on which parameter is being assessed, it may be necessary to collect a sample of water in a clear glass bottle for assessment purposes. The following parameters can be assessed:

- ***Colour:*** Is the water colourless or does it have a colour (e.g. milky, turbid, light brown, dark brown, light blue or green)? Colour in water can be due to pollutants or suspended matter. Look for dramatic changes in water colour when assessing this parameter.
- ***Oil sheen:*** An oil sheen is present if a film of iridescent colour is noted on the surface of the sample. Look for a rainbow effect that can appear to be floating on the surface of the water.
- ***Odour:*** Note whether any odours are present and what they smell like. For example, rotten eggs (hydrogen sulphide), a sour smell, sewage, chlorine, petrol fumes, freshly mown grass or dead fish.
- ***Foam:*** Gently shake the sample and observe any foaming. Foam in the sample is most likely caused by surfactants and may resemble dish-washing soapsuds. Is there an accumulation of foam where water falls over a weir?
- ***Stream bank:*** Is there natural riparian vegetation or is it degraded? Are the river banks collapsed and eroded? Is there garbage next to the river?
- ***Biological characteristics:*** Is there wildlife, such as birds, amphibian, reptiles or mammals, in or around the river? Are there fish in the river? Are there any aquatic plants? Are these aquatic plants attached, limited to the river bank, covering the entire surface of the river? Is there any algae in the river? What does the algae look like (e.g. filamentous algae or mats of algae floating on the water's surface)?



Additional reading material:

- United States Department of Agriculture. Stream Visual Assessment Protocol. www.nrcs.usda.gov/technical/ECS/aquatic/svapfnl.pdf

1.2 RIVER SAMPLING PROCEDURES

1.2.1 General preparation and sampling procedures

Reused sample containers and glassware must be cleaned and rinsed before the first sampling run and after each run. Unless specified in the sampling procedure, the following General Preparation for Sampling Containers can be used.

General Preparation of Sampling Containers
The following method should be used when preparing all sample containers and glassware for chemical analysis, monitoring of turbidity, pH and dissolved oxygen. Wear latex gloves!

1. Wash each sample bottle or piece of glassware with a brush and phosphate-free liquid detergent.

2. Rinse three times with cold tap water.

3. Rinse three times with distilled or deionised water.

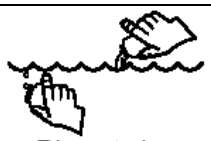
1.2.2 Collecting a sub-surface grab sample in a river

In general, sample away from the stream bank in the main current where the water in the stream has been well mixed. Never sample stagnant water. The outside curve of the stream is often a good place to sample, since the main current tends to hug this bank. In shallow stretches, carefully wade into the centre current to collect the sample. To collect water samples using screw-cap sample bottles, use the following procedures:

1. Label the bottle with the site number, date and time and the name of the sampler.

2. Remove the cap from the bottle just before sampling. Avoid touching the inside of the bottle or the cap. If you accidentally touch the inside of the bottle, use another one.

3. *Wading*: Try to disturb as little bottom sediment as possible. Be careful not to collect water that has sediment from bottom disturbance. Stand facing upstream. Collect the water sample on your upstream side, in front of you. You may also place your bottle to an extension pole to sample from deeper water.

1	• Hold the bottle near its base and plunge it (opening downward) below the water surface, fill with water and discard to rinse. • Repeat this step.	 Rinse twice
2	• Collect a water sample ± 15 cm beneath the surface or mid-way between the surface and the bottom if the stream reach is shallow. • Turn the bottle underwater into the current and away from you. In slow-moving stream reaches, push the bottle underneath the surface and away from you in an upstream direction.	 Take sample
3	Leave a ± 2 cm air space (except for dissolved oxygen samples). Do not fill the bottle completely (so that the sample can be shaken just before analysis). Recap the bottle carefully, remembering not to touch the inside.	 Decant 2 cm

4	• Fill in the bottle number and/or site number on the appropriate field data sheet. • If the samples are to be analyzed in a laboratory, place them on ice in a cooler box for transport to the lab.	 Close tightly

1.2.3 Collecting a surface grab sample

This method is based on the one used by DWA using a bucket to collect the sample. It should not be used when an obvious surface algal scum is present.

Apparatus:

- A clean plastic bucket. If the sample is to be taken from the bank, then a 3 m rope should be attached.
- A clean plastic sample bottle with screw cap. Ensure the bottle is clean and clearly marked (site number, date and time and the name of the sampler).

1	• Throw the bucket out into the water. • Using the rope, haul in across the surface filling the bucket with water. • Rinse and discard contents. Repeat this step.	 Rinse twice
2	Again throw the bucket out into the water and haul in collecting the sample.	 Take sample
3	Fill the sample bottle. Ensure the correctly marked sample bottle is used.	 Fill sample bottle
4	Pour out a small amount (about 2 cm from the top of bottle). This allows the sample to be properly mixed before analysis.	 Decant 2 cm
5	Screw cap tightly into place.	 Close tightly

1.3 TEMPERATURE

Why is temperature important?

Temperature is important because it determines the kinds of aquatic life that can live in the river. Fish, insects, zooplankton, phytoplankton all have a preferred temperature range. If the temperatures are outside of this temperature range for a prolonged period of time, the number of individuals of the species decreases until there are finally none.

Temperature is also important because it influences water chemistry. The rate of chemical reactions generally increases at higher temperatures, which in turn affects biological activity. An example of the effects of temperature on water chemistry is its impact on the oxygen content of the water. Warm water holds less oxygen than cool water, so it may be saturated with oxygen, but still not contain enough oxygen for the survival of aquatic life. Temperature also impacts on the rate of photosynthesis by aquatic plants, the metabolic rates of aquatic organisms and the sensitivity of organisms to toxic wastes, parasites and disease.

What are the reasons for variation in temperature?

In addition to seasonal variations in river temperature caused by changing air temperatures, many other physical aspects of a river also cause natural variation in temperature, including:

- The source of the river;
- Hydrological factors such as the source of water, relative contribution of groundwater and the rate of flow or discharge;
- Climatic factors such as air temperature, cloud cover, wind speed, vapour pressure and rain events;
- Structural characteristic of the river and the catchment area such as shading by riparian vegetation, channel form (depth and width), water volume, turbidity, nature of the stream bank and topographical features.

Man-made sources which results in thermal pollution include:

- Discharge of heated industrial effluents;
- Discharge of heated effluents below power stations;
- Heated return-flows of irrigation water;
- Removal of riparian vegetation cover, thereby exposing the stream to increased warming by sunlight;
- Inter-basin transfers; and
- Discharge of cold bottom water from impoundments (dams).

Sampling and equipment considerations:

As mentioned above, temperature in a stream will vary with width and depth and the time of day. Therefore temperature should always be measured at the same place and at the same time every time. “Spot” or instantaneous measurements can be used to calculate seasonal minimums and maximums.

Temperature is measured in situ using a thermometer and is expressed as degrees Celsius (°C). The procedure for measuring temperature consists of the following tasks:

TASK 1 : PREPARE BEFORE LEAVING FOR THE SAMPLE SITE

Refer to **Part 2** for details on confirming sampling date, time safety considerations, checking supplies and checking weather and directions.

In addition to the standard sampling equipment you will need:

- A thermometer; and
- A data sheet for recording temperature readings.

TASK 2 : MEASURE THE TEMPERATURE

In general, sample away from the stream bank in the main current. In shallow stretches, wade into the centre current to measure temperature. If wading is not possible, tape the thermometer to an extension pole. If you are using an extension pole, read the temperature quickly before it changes to the air temperature. Measure temperature as follows:

1. Place the thermometer in the water at least 30 cm below the surface. Allow enough time for the thermometer to reach a stable temperature (at least 1 minute).

2. If possible, try to read the temperature with the thermometer bulb beneath the water surface. If it is not possible, quickly remove the thermometer and read the temperature.

3. Record the temperature on the field data sheet.

1.4 WATER CLARITY / TURBIDITY

Why is turbidity important?

Turbidity is a measure of water clarity and determines by how much material is suspended in the water. Suspended materials include soil particles (cay slit and sand), algae, plankton, microbes and other substances. Turbidity can also affect the colour of the water. The impacts of high turbidity include:

- Increased sedimentation and siltation in the river, which can reduce available habitats for fish and other aquatic life.
- Increase in the water temperature, as suspended particles absorb more heat. This in turn reduces the concentration of dissolved oxygen, because warm water holds less oxygen than cold water.
- Reduction in the amount of light penetrating the water, which reduces the rate of photosynthesis and the production of dissolved oxygen.
- Fish kills as suspended materials may clog fish gills, reducing the resistance to disease in fish, lowering growth rates and affecting egg and larval development.

What are the sources of suspended materials (i.e. turbidity)

Land use is probably the greatest factor influencing changes in turbidity in rivers. As catchment areas develop, there is an increase in disturbed areas and a decrease in vegetation, which in turn increases the rate of soil erosion. Turbidity is therefore a useful indicator of the effects of runoff from construction, agricultural practices, discharges and other land use practices. Other sources of turbidity include:

- Soil erosion;
- Waste discharge;
- Urban runoff;
- Eroding stream banks;
- Large numbers of bottom feeders (such as carp), which stir up bottom sediments; and
- Excessive algal growth.

Sampling and equipment considerations:

Turbidity is not a direct measure of the mass of suspended solids present, although the two are related, or the rate of sedimentation of a river, it is only a measure of the clarity of the river. Turbidity measurements are reported in nephelometric turbidity units (NTU).In a river, turbidity is best measured using a transparency tube. The turbidity tube uses the correlation between visibility and turbidity to approximate a turbidity level (i.e. the higher the turbidity, the harder it is to see through the water).

The key components of the turbidity tube are: a clear tube, tube cap, viewing disc and a measuring device (ruler). A description of how to assemble the turbidity tube is provided in **Appendix 2**. The viewing disk should be placed in the tube cap which is used to seal the tube. The water sample is then decanted into the tube until the viewing disk can no longer be seen. The height from which the marker can no longer be seen correlates to a known turbidity value. **Table 1.1** provides the turbidity value (in NTU) that corresponds to different heights measured above the viewing disk.

Table 1.1: Water height-to-turbidity conversion

Centimetres	NTU
6.7	240
7.3	200
8.9	150
11.5	100
17.9	50
20.4	40
25.5	30
33.1	21
35.6	19
38.2	17
40.7	15
43.3	14
45.8	13
48.3	12
50.9	11
53.4	10
85.4	5

TASK 1 : PREPARE BEFORE LEAVING FOR THE SAMPLE SITE

Refer to **Part 2** for details on confirming sampling date, time safety considerations, checking supplies and checking weather and directions. In addition to the standard sampling equipment you will need:

- A turbidity tube;
- Clean bucket or container to collect water samples; and
- A data sheet for recording temperature readings.

TASK 2 : TAKING THE MEASUREMENT USING A TURBIDITY TUBE

Before you begin:

1. Measurements should be taken in daylight, but not direct sunlight. Cast a shadow on the tube by placing yourself between the sun and the tube.
2. Do not wear sunglasses when reading the tube!

Measurement procedure:

1. Dip the container/bucket into the water to collect the sample. Be careful not to include sediment from the bottom of the river. Rinse the tube with the water that is going to be tested and pour it out.

2. Stir or swirl the water sample in the container vigorously until it is homogenous, introducing as little air as possible.

3. Place your head 10 – 20 cm directly over the tube so that you can see the viewing disk while the sample is being poured into the tube (steps 4 – 8 are illustrated in **Figure 1.1**).

4. Slowly pour water into the tube. Try not to form bubbles as your pour. If bubbles do form: stop pouring and allow any bubbles to rise and the surface of the water to become still.

5. Keep slowly adding water until the pattern on the disk becomes hard to see. Stop pouring as soon as the pattern on the disk can no longer be seen.

6. Read the height of water in the turbidity tube and find the corresponding turbidity value in **Table 1.1**. If you can still see the viewing disk pattern when the tube is full: record the turbidity value as greater than the final measuring mark (or less than 5 NTU).

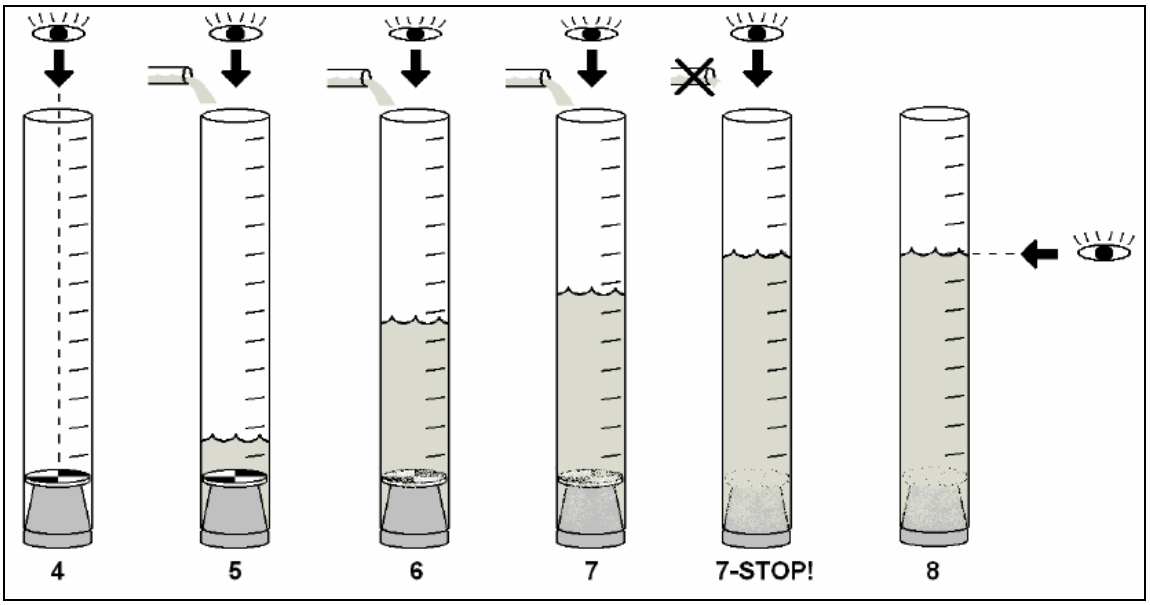


Figure 1.2: Measurement procedure for the turbidity tube (Myre and Shaw, 2006)

1.5 DISSOLVED OXYGEN

Why is dissolved oxygen important?

Like all terrestrial animals, fish and other aquatic organisms need oxygen to live. In rivers, oxygen is dissolved in water and is either lost or gained through various processes. Dissolved oxygen (DO) is gained from the atmosphere and from plants as a result of photosynthesis. While DO is lost through respiration by aquatic animals and plants, decomposition and various chemical reactions. A certain minimum amount of DO must be present in the water for aquatic life to be sustained.

Why does the level of DO vary?

Oxygen is produced during photosynthesis and consumed during respiration and decomposition. Because photosynthesis requires light, oxygen is only produced by plants during the daylight hours. Decomposition and respiration are not limited by the availability of light and occur 24-hours a day. During the night, when there is no photosynthesis, the DO concentrations steadily decline. They are lowest just before dawn, when photosynthesis resumes. This difference in the production and consumption of oxygen can account for the large daily variation in DO levels. This is called the diurnal variation in DO.

DO concentration increases wherever water flows becomes turbulent, such as in a rapid or below a waterfall. Another physical process which also impacts on DO concentrations is the temperature of the water. Cold water can hold more DO than warm water. So during the summer months, when the river is warmer, the DO concentration can be limited by the ability of the water to “soak up” more oxygen (**Table 1.1**). It is therefore important to note the time of your DO sampling to help judge when in the daily cycle the data was collected.

Table 1.2: Maximum DO concentrations showing temperature variation

Temperature (°C)	DO (mg/l)	Temperature (°C)	DO (mg/l)
0	14.60	23	8.56
1	14.19	24	8.40
2	13.81	25	8.24
3	13.44	26	8.09
4	13.09	27	7.95
5	12.75	28	7.81
6	12.43	29	7.67
7	12.12	30	7.54
8	11.83	31	7.41
9	11.55	32	7.28
10	11.27	33	7.16
11	11.01	34	7.16
12	10.76	35	6.93
13	10.52	36	6.82
14	10.29	37	6.71
15	10.07	38	6.61
16	9.85	39	6.51
17	9.65	40	6.41
18	9.45	41	6.41
19	9.26	42	6.22
20	9.07	43	6.13
21	8.90	44	6.04

Temperature (°C)	DO (mg/l)	Temperature (°C)	DO (mg/l)
22	8.72	45	5.95

Pollution tends to cause a decrease in DO levels. This can be caused by the addition of effluent or run-off water with a low concentration of DO or chemical or biological constituents that have a high oxygen demand (i.e. they require large amounts of oxygen for before they can be decomposed). The amount of oxygen consumed by these organisms which decompose the chemical or biological constituents is known as the biochemical oxygen demand (BOD).

Sampling and equipment considerations:

In rivers, the DO levels are most likely to vary horizontally along the course of the waterway. The DO levels in and below rapids and waterfalls, are typically higher than those in pools and slower moving stretches.

Dissolved oxygen is measured either by using some variation of the Winkler method (chemical method) or by using a dissolved oxygen meter and probe. The DO measurement is either in milligrams per litre (mg/l) or "percent saturation." DO measured in mg/l provides an indication of the amount of oxygen in a litre of water. Percent saturation is the amount of oxygen in a litre of water relative to the total amount of oxygen that the water can hold at that temperature.

DO samples are collected using a special BOD bottle: a glass bottle with a "turtleneck" and a ground glass stopper. You can fill the bottle directly in the stream if the stream is wadable or boatable, or you can use a sampler that is dropped from a bridge or boat into water deep enough to submerse the sampler. Samplers can be made or purchased.

The Winkler method involves filling a sample bottle completely with water (no air is left to bias the test). The dissolved oxygen is then "fixed" using a series of chemical reagents that form an acid compound that is titrated. Titration involves the drop-by-drop addition of a reagent that neutralizes the acid compound and causes a change in the colour of the solution. The point at which the colour changes is the "endpoint" and is equivalent to the amount of oxygen dissolved in the sample. The sample is usually fixed and titrated in the field at the sample site. It is possible, however, to prepare the sample in the field and deliver it to a lab for titration.

A dissolved oxygen meter is an electronic device that converts signals from a probe that is placed in the water into units of DO in mg/l. Most meters and probes also measure temperature. The probe is filled with a salt solution and has a selectively permeable membrane that allows DO to pass from the stream water into the salt solution. The DO that has diffused into the salt solution changes the electric potential of the salt solution and to mg/l.

TASK 1 : PREPARE BEFORE LEAVING FOR THE SAMPLE SITE

Refer to **Part 2** for details on confirming sampling date, time safety considerations, checking supplies and checking weather and directions. In addition to the standard sampling equipment you will need:

If using the Winkler Method:

- Labels for sample bottles;
- Field kit and instructions for DO testing;
- Enough reagents for the number of sites to be tested;
- Kemmerer, Van Doorn or home-made sampler to collect deep-water samples;
- A numbered glass BOD bottle with a glass stopper (one for each site); and

- Data sheets to record DO results.

If using a DO meter and probe:

- DO meter and probe (electrode) (NOTE: Confirm that the meter has been calibrated according to the manufacturer's instructions);
- Operating manual for the meter and probe;
- Extra membranes and electrolyte solution for the probe;
- Extra batteries for the meter;
- Extension pole; and
- Data sheet to recode DO results.

TASK 2 : COLLECT SAMPLES

Winkler method procedure:
Use a BOD bottle to collect the water sample. The most common sizes are 300 ml and 60 ml. Be sure that you are using the correct volume for the titration method that will be used to determine the amount of DO. Use the following procedure for collecting a sample for titration by the Winkler method:

Remember that the water sample must be collected in such a way that you can cap the bottle while it is still submerged. That means that you must be able to reach into the water with both arms and the water must be deeper than the sample bottle.

1. Carefully wade into the stream. Stand so that you are facing one of the banks.

2. Collect the sample so that you are not standing upstream of the bottle. Remove the cap of the BOD bottle. Slowly lower the bottle into the water, pointing it downstream, until the lower lip of the opening is just submerged. Allow the water to fill the bottle very gradually, avoiding any turbulence (which would add oxygen to the sample). When the water level in the bottle has stabilized (it won't be full because the bottle is tilted), slowly turn the bottle upright and fill it completely. Keep the bottle under water and allow it to overflow for 2 or 3 minutes to ensure that no air bubbles are trapped.

3. Cap the bottle while it is still submerged. Lift it out of the water and look around the "collar" of the bottle just below the bottom of the stopper. If you see an air bubble, pour out the sample and try again.

4. “Fix” the sample immediately following the directions in your kit:

    a. Remove the stopper and add the fixing reagents to the sample.

    b. Immediately insert the stopper so air is not trapped in the bottle and invert several times to mix. This solution is caustic. Rinse your hands if you get any solution on them. An orange-brown flocculent precipitate will form if oxygen is present.

    c. Wait a few minutes until the floc in the solution has settled. Again invert the bottle several times and wait until the floc has settled. This ensures complete reaction of the sample and reagents. The sample is now fixed, and atmospheric oxygen can no longer affect it. If you are taking the sample to the lab for titration, no further action is necessary. You can store the sample in a cooler for up to 8 hours before

titrating it in a laboratory. If you are titrating the sample in the field, see **Task 3: Analyze the Samples**.

Using a DO meter and probe:
If you are using a dissolved oxygen meter, be sure that it is calibrated immediately prior to use. Check the cable connection between the probe and the meter. Make sure that the probe is filled with electrolyte solution, that the membrane has no wrinkles, and that there are no bubbles trapped on the face of the membrane. You can do a field check of the meter's accuracy by calibrating it in saturated air according to the manufacturer's instructions. Or, you can measure a water sample that is saturated with oxygen, as follows. (NOTE: You can also use this procedure for testing the accuracy of the Winkler method.)

1. Fill a 1-liter beaker or bucket of tap water. Mark the bottle number as "tap" on the lab sheet.

2. Pour this water back and forth into another beaker 10 times to saturate the water with oxygen.

3. Use the meter to measure the water temperature and record it in the water temperature column on the field data sheet.

4. Find the water temperature of your "tap" sample in **Table 1.1**. Use the meter to compare the dissolved oxygen concentration of your sample with the maximum concentration at that temperature in the table. Your sample should be within 0.5 mg/l. If it is not, repeat the check and if there is still an error, check the meter's batteries and follow the troubleshooting procedures in the manufacturer's manual.

Once the meter is turned on, allow for the specified time in the manual before calibrating. After calibration, do not turn the meter off until the sample is analyzed. Once you have verified that the meter is working properly, you are ready to measure the DO levels at the sampling site. You might need an extension pole (this can be as simple as a piece of wood) to get the probe to the proper sampling point. Simply secure the probe to the end of the extension pole. To use the probe, proceed as follows:

1. Place the probe in the stream below the surface.

2. Set the meter to measure temperature, and allow the temperature reading to stabilize. Record the temperature on the field data sheet.

3. Switch the meter to read dissolved oxygen.

4. Record the dissolved oxygen level on the field data sheet.

TASK 3 : ANALYSING THE SAMPLES
Three types of titration apparatus can be used with the Winkler method: droppers, digital titrators and burets. The dropper and digital titrator are suited for field use. The buret is more conveniently used in the laboratory. Volunteer programs are most likely to use the dropper or digital titrator. For titration with a dropper or syringe, which is relatively simple, follow the manufacturer's instructions. The following procedure is for using a digital titrator to determine the quantity of dissolved oxygen in a fixed sample:

1. Select a sample volume and sodium thiosulfate titration cartridge for the digital titrator corresponding to the expected dissolved oxygen concentration according to **Table 1.1**. In most cases, you will use the 0.2 N cartridge and the 100 ml sample volume.

2. Insert a clean delivery tube into the titration cartridge.

3. Attach the cartridge to the titrator body.

4. Hold the titrator with the cartridge tip up. Turn the delivery knob to eject air and a few drops of titrant. Reset the counter to 0 and wipe the tip.

5. Use a graduated cylinder to measure the sample volume (from the "fixed" sample in the 300 ml BOD bottle) according to **Table 1.2**.

6. Transfer the sample into a 250 ml Erlenmeyer flask, and place the flask on a magnetic stirrer with a stir bar. If you are in the field, you can manually swirl the flask to mix.

7. Place the delivery tube tip into the solution and turn the stirrer on to stir the sample while you're turning the delivery knob.

8. Titrate to a pale yellow colour.

9. Add two dropperfuls of starch indicator solution and swirl to mix. A strong blue colour will develop.

10. Continue to titrate until the sample is clear. Record the number of digits required. (The colour might reappear after standing a few minutes, but this is not a cause for concern. The "first" disappearance of the blue colour is considered the endpoint.)

11. Calculate mg/l of DO = digits required X digit multiplier (from **Table 1.2**).

12. Record the results in the appropriate column of the data sheet.

Table 1.3: Sample volume selection and corresponding values for Winkler titration

Expected range (mg/l)	Sample volume (ml)	Titration cartridge	Digit multiplier
1 – 5	200	0.2 N	0.01
2 – 10	100	0.2 N	0.02
10 +	200	2.0 N	0.10

Some water quality standards are expressed in terms of percent saturation. To calculate percent saturation of the sample:

1. Find the temperature of your water sample as measured in the field.

2. Find the maximum concentration of your sample at that temperature as given in **Table 1.1**.

3. Calculate the percent saturation, by dividing your actual dissolved oxygen by the maximum concentration at the sample temperature.

4. Record the percent saturation in the appropriate column on the data sheet.

1.6 SAMPLING FOR CHEMICAL ANALYSIS

Water contains a host of constituents including:

- Physical soil and clay particles and organic matter from storm runoff;
- Microorganisms, such as bacteria, viruses and parasites;
- Chemical constituents, which can be subdivided into (a) major inorganic chemical salts (such as sodium, chloride, calcium, sulphate, etc.), (b) minor inorganic chemical salts (such as ammonia, fluoride, phosphate and trace metals such as iron, manganese, copper, etc.) and (c) organic substances such as pesticide residues; and
- Radioactive substances (which usually occur only in minute concentrations under natural conditions).

These constituents, and their respective concentration within the water resource, determine the fitness for a variety of uses (domestic, recreation, agriculture and industry) and for the protection of the health and integrity of aquatic ecosystems.

The chemical composition of a water resource is a function of the catchment geology and climate. Human activities, such as the discharge of effluent from mines, wastewater treatment works and industries and run-off from agricultural land, also play a big role in determining the chemical constituents of the water resource. A typical freshwater resource has a balance of three major anions (negatively charged constituents) and four major cations (positively charged constituents).

Table 1.4: Typical freshwater chemical constituents

Anions		Cations	
Carbonate	HCO_3^-	Calcium	Ca^{+2}
Sulphate	SO_4^{-2}	Magnesium	Mg^{+2}
Chloride	Cl^-	Sodium	Na^+
		Potassium	K^+

Sampling and equipment considerations:
Chemical analysis is generally done in a laboratory. The ions most commonly found in natural water resources are provided in **Table 1.4**. The results provide an indication of the ion balance and the inorganic chemical water quality of a water resource.

TASK 1 : PREPARE BEFORE LEAVING FOR THE SAMPLE SITE
Refer to **Part 2** for details on confirming sampling date, time safety considerations, checking supplies and checking weather and directions. In addition to the standard sampling equipment you will need:

- Labels for sample bottles;
- Clean sample bottles, which have been washed in special phosphate free cleaning agents; and
- If the DWAF water laboratory is going to do the analysis, they will supply ampoules of mercury chloride, which is a preservative used to fix the sample.

TASK 2 : COLLECT SAMPLES
Collecting samples from a river without a gauging structure:

- Never sample on one side of an island. Rather collect the sample at a central point in the main stream.

- When sampling downstream of a confluence, remember to select a point where water from both part of the stream are thoroughly mixed.

Collecting samples from a river with a measuring / gauging structure:

- The ideal place to sample is directly below the weir where water flows through.
- When the weir is flooded it is safer to collect the sample downstream from the weir where water is still thoroughly mixed.

Sampling procedure:

- Leave the lid on the bottle until you are ready to take the sample.
- Collect a little bit of water in the sample bottle and rise the bottle. Pour the water out, away from where the sample will be taken.
- Collect the sample by lowering the bottle into the water. Perform a “forward scoop” motion to collect the sample. Do not fill the bottle right up to the top.
- If DWA is the analysing laboratory, then preserve the sample with one ampoule containing mercury chloride. Break the ampoule and the drop both pieces into the sample bottle.
- Label the sample bottle.
- Store the sample bottle in a cooler box or dark container.
- Transport the sample to the laboratory without delay.

1.7 SAMPLING FOR BACTERIOLOGICAL ANALYSIS

Why are bacteria important?

In order to determine where the river has been contaminated by sewage, members of two bacteria groups that are commonly found in human and animal faeces, coliforms and fecal streptococci, are used as indicators. Although these bacteria are generally not harmful themselves, they indicate the possible presence of pathogenic (disease-causing) bacteria, viruses and protozoa that also live in human and animal digestive systems. Therefore, their presence in streams suggests that pathogenic micro-organisms might also be present and that swimming and eating shellfish might be a health risk.

Since it is difficult, time-consuming and expensive to test directly for the presence of a large variety of pathogens, water is usually tested for indicator organisms instead. Sources of faecal contamination to surface waters include wastewater treatment plants, on-site septic systems, domestic and wild animal manure and storm runoff. In addition to the possible health risk associated with the presence of elevated levels of faecal bacteria, they can also cause cloudy water, unpleasant odours and an increased oxygen demand.

The most commonly tested faecal bacteria indicators include total coliforms, faecal coliforms, *Escherichia coli* and enterococci. Each indicator bacteria is used for specific monitoring purposes, which are described below.

- *Total coliform bacteria*: Primarily used as a practical indicator of the general hygienic quality of water. Mainly used in routine monitoring of drinking water supplies.
- *Faecal coliform bacteria*: Primarily used as a practical indicator of faecal pollution, more specific for faecal pollution than total coliforms. Mainly used for assessment of faecal pollution of wastewater, raw water supplies and natural water environments used for recreational purposes.
- *Escherichia coli*: Highly specific indicator of faecal pollution which originates from humans and warm-blooded animals.
- *Enterococci* (*faecal streptococci*): Relatively specific indicators of faecal pollution which tend to survive longer in water environments than coliform bacteria.

Sampling and equipment considerations:

Bacteria can be difficult to sample and analyse, for many reasons including:

- Natural variation of bacteria levels;
- Weather conditions, bacteria levels are strongly correlated with rainfall, and thus comparing wet and dry weather bacteria data can be a problem;
- Many analytical methods have a low level of precision yet can be quite complex; and
- Absolutely sterile conditions are required to collect and handle samples.

The primary equipment decision to make when sampling for bacteria is what type and size of sample container you will use. Once you have made that decision, the same, straightforward collection procedure is used regardless of the type of bacteria being monitored. It is critical when monitoring bacteria that all containers and surfaces with which the sample will come into contact be sterile. Containers made of either some form of plastic or Pyrex glass are acceptable. However, if the containers are to be reused, they must be sterilized using heat and pressure. Plastic containers, made from either high-density polyethylene or polypropylene, might be preferable to glass from a practical standpoint because they will better withstand breakage. The size of the container will depend on the sample amount

needed for the bacteria analysis method you choose and the amount needed for other analyses. There are two basic methods for analysing water samples for bacteria:

1. The membrane filtration method involves filtering several different-sized portions of the sample using filters with a standard diameter and pore size, placing each filter on a selective nutrient medium in a petri plate, incubating the plates at a specified temperature for a specified time period, and then counting the colonies that have grown on the filter. This method varies for different bacteria types (variations might include, for example, the nutrient medium type, the number and types of incubations, etc.).

2. The multiple-tube fermentation method involves adding specified quantities of the sample to tubes containing a nutrient broth, incubating the tubes at a specified temperature for a specified time period, and then looking for the development of gas and/or turbidity that the bacteria produce. The presence or absence of gas in each tube is used to calculate an index known as the Most Probable Number (MPN).

Given the complexity of the analysis procedures and the equipment required, field analysis of bacteria is not recommended.

TASK 1 : PREPARE BEFORE LEAVING FOR THE SAMPLE SITE

Refer to **Part 2** for details on confirming sampling date, time safety considerations, checking supplies and checking weather and directions. In addition to the standard sampling equipment you will need:

1. Labels for sample bottles; and
2. Sterilised sample bottles.

TASK 2 : COLLECT SAMPLES

Samples should be collected using the standard procedure described in **Part 2**. Note that sample bottles must be sterilised before they are used. Be careful not to touch the inside of the bottle or the lid.

Remember to wash your hands thoroughly after collecting samples suspected of containing faecal contamination. Also be careful not to touch your eyes, ear, nose or mouth until your have washed your hands.

Samples for bacteria must be analysed within six hours of collection. Keep the samples on ice and take them to the laboratory as soon as possible.

1.8 PH

What is pH and why is it important?

The pH of a sample is a terms used to indicate the alkalinity or acidity of a substance ranked on a scale from 1.0 to 14.0. A pH of 7 is considered to be neutral. Substances with pH less than 7 are acidic, while substances with pH greater than 7 are basic. The pH of most natural waters ranges between 6.5 and 8.5. **Figure 1.3** presents the pH of some of the common liquids.

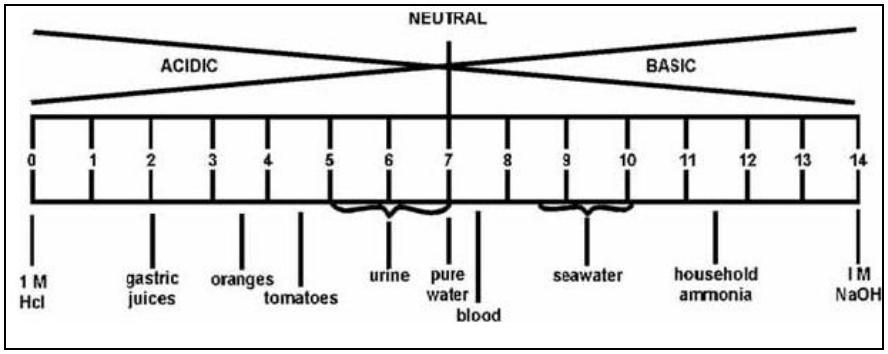


Figure 1.3: pH scale

The pH of water determines the solubility (amount that can be dissolved in the water) and biological availability (amount that can be utilised by aquatic life) of chemical constituents, such as nutrients (e.g. phosphorus, nitrogen and carbon) and heavy metals (e.g. lead, cadmium and copper). For example, pH can determine how much and in what form phosphorus is most abundant in the water. pH can also allow toxic elements and compounds to become mobile and "available" for uptake by aquatic animals and plants. pH also affects biological processes in water. For example, different organisms flourish within different ranges of pH. The largest variety of aquatic organisms prefer a pH range between 6.5 and 8.0. pH outside this range reduces the diversity in the river because it stresses the organisms and can reduce reproduction.

The pH scale measures the logarithmic concentration of hydrogen (H^+) and hydroxide (OH^-) ions, which make up water ($H^+ + OH^- = H_2O$). When both types of ions are in equal concentration, the pH is 7.0 or neutral. Below 7.0, the water is acidic (there are more hydrogen ions than hydroxide ions). When the pH is above 7.0, the water is alkaline, or basic (there are more hydroxide ions than hydrogen ions). Since the scale is logarithmic, a drop in the pH by 1.0 unit is equivalent to a 10-fold increase in acidity. So, a water sample with a pH of 5.0 is 10 times as acidic as one with a pH of 6.0, and pH 4.0 is 100 times as acidic as pH 6.0.

Why does the pH vary?

The geology of the catchment and the original source of the water determines the initial pH of the water. The greatest natural cause for change in pH is the seasonal and daily variation in photosynthesis. Photosynthesis uses up hydrogen molecules, which causes the concentration of H+ ions to decrease and therefore the pH increases. Respiration and decomposition processes lower the pH. For this reason pH is higher during daylight hours and during the growing seasons when photosynthesis is at its peak. Although pH may constantly change, the amount of change is remains within a narrow pH range. Natural waters also have the ability to prevent major changes in pH. Small or localised changes in pH are quickly modified by various chemical reactions so little or no change may be measures. The ability to resist change in pH is called "buffering capacity".

Industrial activities generally cause acidification rather than alkalinisation of rivers. Acidification is normally the result of three different types of pollution, namely:

- Low pH point-source effluents from industries, such as pulp and paper and tanning and leather industries;
- Mine drainage, which is nearly always acid, leading to the pH of receiving streams dropping to below two; and
- Acid precipitation resulting largely from atmospheric pollution caused by the burning of coal (and subsequent production of sulphur dioxide (SO_2)) and the exhausts of combustion engines (nitrogen oxides (NO_x)). Both sulphur oxides (SO_x) and nitrogen oxides (NO_x) form strong mineral acids when dissolved in water. When acid rain falls on a catchment, the strong acids leach calcium and magnesium from the soil and also interfere with nutrient availability.

Elevated pH values can be caused by increased biological activity in eutrophic systems. The pH values may fluctuate widely from below six above ten over a 24-hour period as a result of changing rates of photosynthesis and respiration.

Sampling and equipment considerations:
pH can be analyzed in the field or in the lab. If it is analyzed in the lab, you must measure the pH within two hours of the sample collection. This is because the pH will change due to the carbon dioxide from the air dissolving in the water, which will bring the pH toward seven. Alternatively, pH measurements can be taken in the field using pH meters, colour comparators or pH "pocket pals".

pH Meters
A pH meter measures the electric potential (millivolts) across an electrode when immersed in water. This electric potential is a function of the hydrogen ion activity in the sample. Therefore, pH meters can display results in either millivolts (mV) or pH units. A pH meter consists of a potentiometer, which measures electric current; a glass electrode, which senses the electric potential where it meets the water sample; a reference electrode, which provides a constant electric potential; and a temperature compensating device, which adjusts the readings according to the temperature of the sample (since pH varies with temperature). The reference and glass electrodes are frequently combined into a single probe called a combination electrode. There is a wide variety of meters, but the most important part of the pH meter is the electrode.

pH "Pocket Pals" and Colour Comparators
pH "pocket pals" are electronic hand-held "pens" that are dipped in the water and provide a digital readout of the pH. They can be calibrated to one pH buffer. Colour comparators involve adding a reagent to the sample that colours the sample water. The intensity of the colour is proportional to the pH of the sample. This colour is then matched against a standard colour chart. The colour chart equates particular colours to associated pH values. The pH can be determined by matching the colours from the chart to the colour of the sample.

TASK 1 : PREPARE SAMPLE CONTAINERS
Sample containers must be cleaned and rinsed before the first run and after each sampling run by following the procedure described in **Part 2**.

TASK 2 : PREPARE BEFORE LEAVING FOR THE SAMPLING SITE
Refer to **Part 2** for details on confirming sampling date, time safety considerations, checking supplies and checking weather and directions.

In addition to the standard sampling equipment you will need:

- pH meter with combination temperature and reference electrode, or pH "pocket pal" or colour comparator;
- Wash bottle with deionised water to rinse pH meter electrode (if appropriate); and
- Data sheet for pH to record results.

Before you leave for the sampling site, be sure to calibrate the pH meter or "pocket pal." The pH meter and "pocket pal" should be calibrated prior to sample analysis and after every 25 samples according to the instructions that come with them.

TASK 3 : COLLECT THE SAMPLE

Samples are collected using the standard sampling process described in **Part 2**. Which sampling procedure should be used?

TASK 4 : MEASURE pH

The procedure for measuring pH is the same whether it is conducted in the field or lab. If you are using a "pocket pal" or colour comparator, follow the manufacturer's instructions. Use the following steps to determine the pH of your sample if you are using a meter.

1. Rinse the electrode well with deionised water.

2. Place the pH meter or electrode into the sample. Depress the dispenser button once to dispense electrolyte.

3. Read and record the temperature and pH in the appropriate column on the data sheet. Rinse the electrode well with deionised water.

4. Measure the pH of the 4.01 and 7.0 buffers periodically to ensure that the meter is not drifting off calibration. If it has drifted, recalibrate it.

1.9 RIVER HEALTH

Introduction

The health of a river can be measured by its plants and organisms (Palmer *et al*., 2002). A river's health is assessed by how far away it is from the way it would have been naturally, when the impact of people was very low. The assessment asks, for each stretch of the river, how close to natural are the patterns of flow, the kinds of plants and animals, the shape and structure of the ecosystem, and the chemistry of the water.

From these measurements we can work out the class of river health (Palmer *et al*., 2002). ***Natural systems*** are, of course, still natural. ***Good systems*** are slightly changed from natural but still have most of the plants and animals they used to have, including sensitive ones. A few sensitive species may have disappeared, but there is a good natural diversity, and all the natural processes function well. Some water can be or is being used for domestic supply, and some sewage waste is discharged. ***Fair systems*** are "hanging in there". They have lost their sensitive species but are well populated by tough organisms which keep the basic processes going. They provide people with water for domestic use and economic growth and their waste water is transported and diluted. ***Poor rivers*** have lost many species and many functions – they need help. They have had too much water taken out and too many wastes put in. They may be a health hazard.

A variety of invertebrate organisms (snails, crabs, worms, insect larvae and adults, mussels) require specific habitat types and water quality and flow conditions for at least part of their life cycles (Palmer *et al*, 2004). Changes in the structure of invertebrate communities are a sign of changes in overall river conditions, because most invertebrate species are fairly short-lived and remain in one area during their aquatic life phase. This makes them particularly good indicators of localised conditions in a river over the short term. The South African Scoring System (SASS) was based on the presence of families of aquatic invertebrates and their sensitivity to water quality changes. It has been widely tested and used in South Africa as a biological index of water quality.

Measuring the health of a river

MiniSASS is a simple technique that can be used to measure the health of a river and the general quality of the water in that river. It uses the composition of invertebrates (bottom dwelling insects) living in rivers and is based on the sensitivity of the various animals to water quality. It does not, however, measure contamination of the water by bacteria and viruses and thus does not determine if the water is fit to drink without treatment.

The Mini-SASS is a simpler version of the more sophisticated SASS method that is used as part of the National River Health Programme. The results produced using Mini-SASS have been tested against the more rigorous SASS method and have been found to be sufficiently close to be of real value to volunteer monitoring.

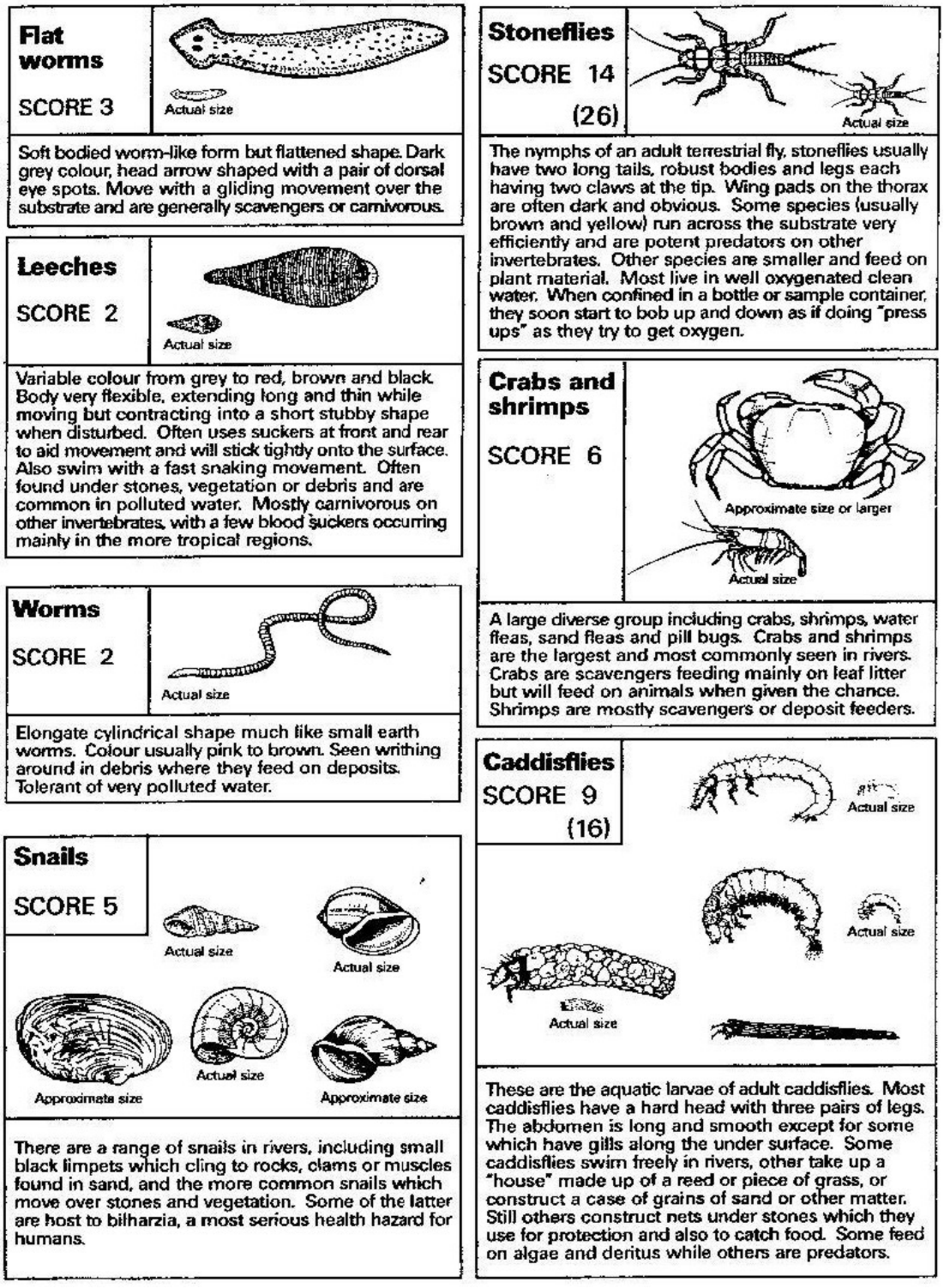
Method:

1. The best sites to find insects in a nearby river are where the current is fairly fast moving and where there is some vegetation growing in the water, along the sides of the river.

2. Look for invertebrates in as many of the different habitats (biotopes) you can find at a river site. Insects are collected holding a small net (a kitchen sieve will do) in the current, and then disturbing the stones, vegetation and sand using your feet (with boots on!) or hands just upstream of the net. Be bold in turning the stones over. The insects will be dislodged

and will flow into the net. Do this for about 5 minutes while ranging across the river to a number of different habitats. You can also lift stones and pick off the insects with your fingers or you can brush off the underside of the stones with a clean paintbrush.

3. Rinse any mud out of the net then turn the contents into a plastic tray (a 2 litre ice-cream container is ideal). Identify each group using the miniSASS sheet (keep a tally of the number of each group). If the river is in reasonable condition, you should have several hundred individual insects in the sample.



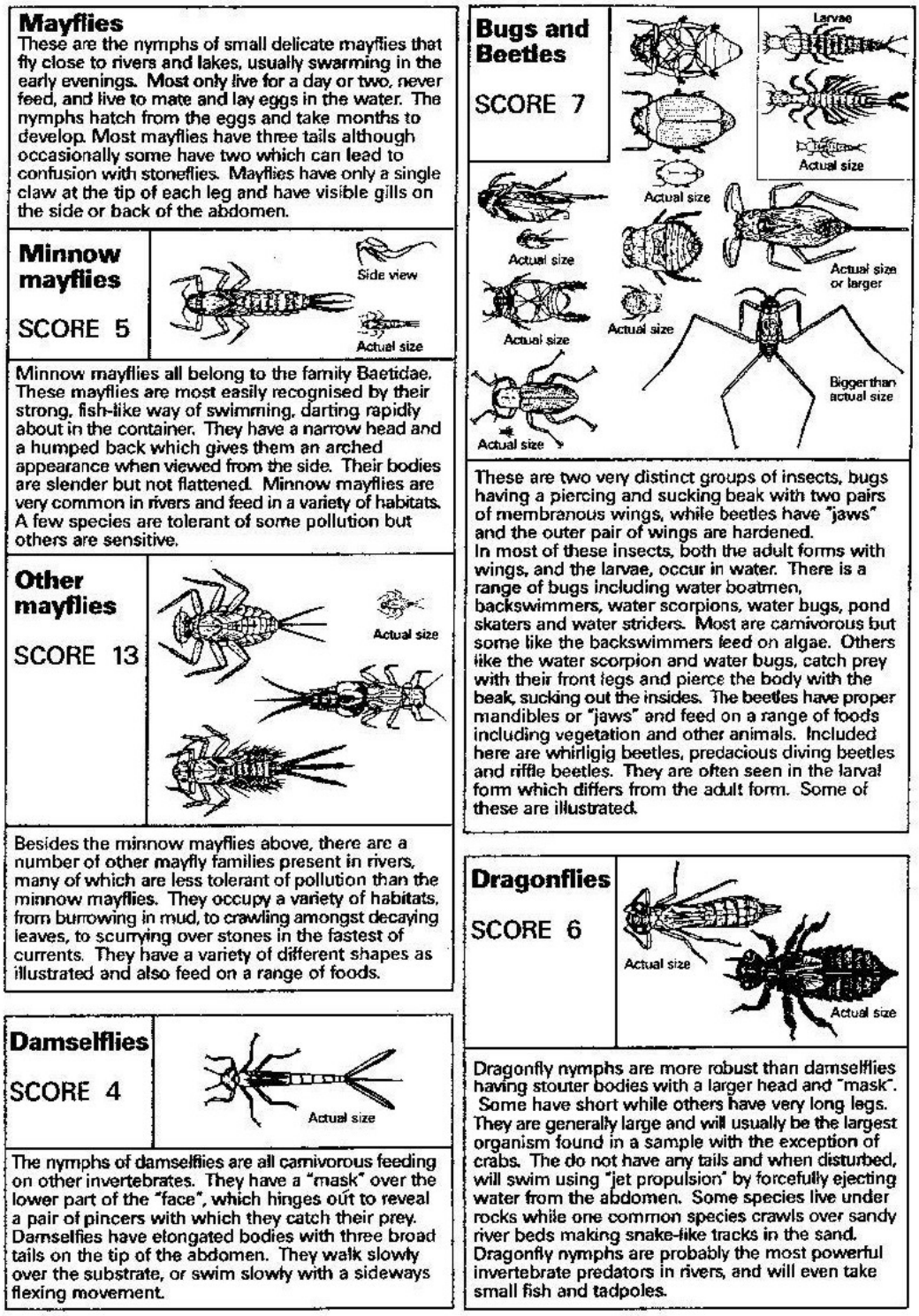


Figure 1.4: Example of the miniSASS sheet for identifying insect groups.

Calculating your river's MiniSASS score

1. For each of the groups found in your sample, circle the score on the miniSASS table.

2. Add up the scores and divide by the number of groups found. This will give you an average score. MiniSASS produces a single score which is similar and comparable to the average score which is produced by the more complex version of SASS.

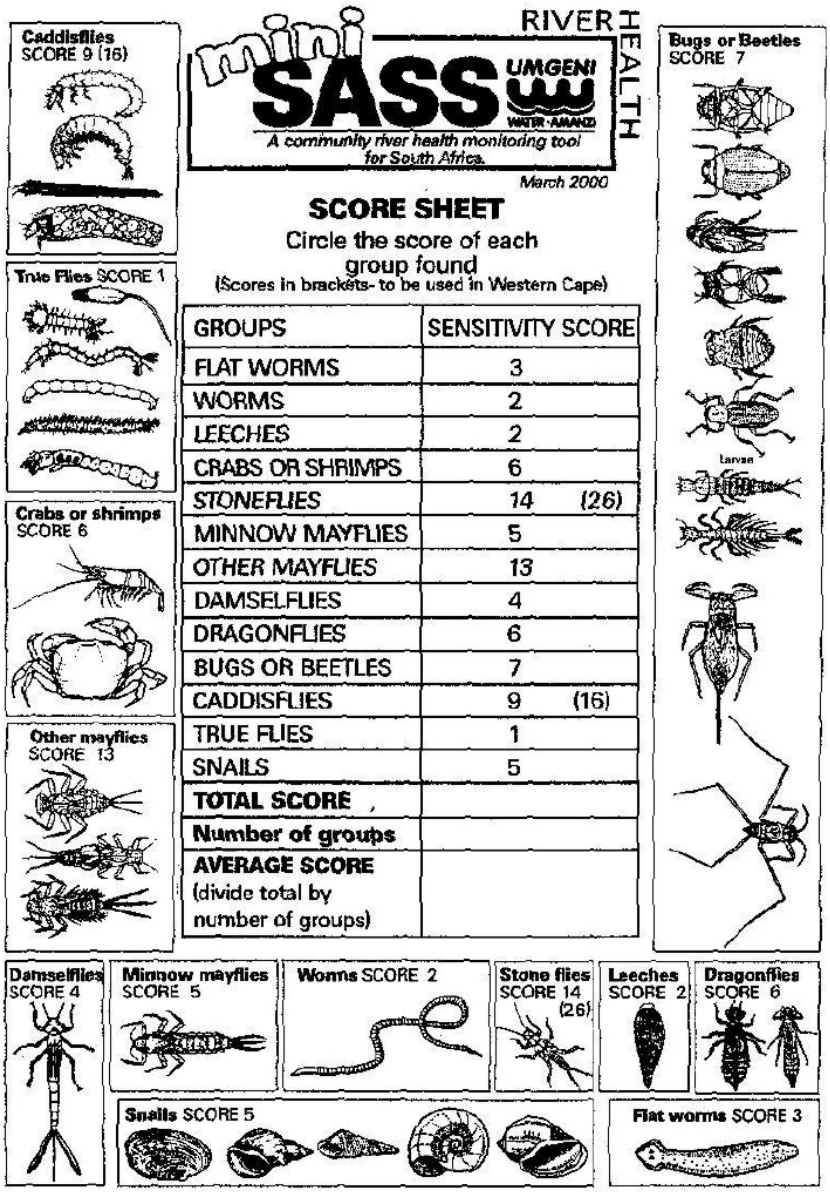


Figure 1.5: Example of the miniSASS score sheet

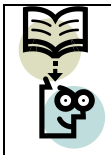
Score	Interpretation
0 - 2	Highly impacted stream (poor condition)
2 - 4	Impacted stream (fair condition)
4 - 6	Slightly impacted stream (good condition)
> 6	Good quality stream (probably approaching natural condition)

On rare occasions, an incorrect result will be obtained when the average score is high but the sample only contained a few (one to three) insect groups. When this happens this means that the river is impacted or disturbed but in a way that favours some organisms.

Remember to return all the insects back to the river.

NOTE: The miniSASS method described here should be replaced by the updated miniSASS methodology that is currently being completed as part of a Water Research Commission project by the University of KwaZulu-Natal and GroundTruth Ecosystem and Environmental Consultants. It is due for completion in 2009.

2. WATER QUALITY MONITORING IN RESERVOIRS



Additional reading material:

- United States Environmental Protection Agency. Volunteer Lake Monitoring: A Methods Manual. http://www.epa.gov/owow/monitoring/lakevm.html
- DWAF: Resource Quality Services. 2004. Water Resource Quality Monitoring. Volume 1: Sampling Protocol for Inorganic Chemical Analysis.
- DWAF: Resource Quality Services. 2004. Water Resource Quality Monitoring. Volume 2: Eutrophication Monitoring.
- Gerber, A., Cilliers, C.J., van Ginkel, C. and Glen, R. 2004. Easy Identification of Aquatic Plants.
- Waterwatch Australia. Module 2: Getting Started: the team, monitoring and site. http://www.waterwatch.org.au/publications/module2/index.html
- Waterwatch Australia. Module 3: Biological Parameters. http://www.waterwatch.org.au/publications/module3/index.html
- Waterwatch Australia. Module 4: Physical and Chemical Parameters. http://www.waterwatch.org.au/publications/module4/index.html

The following water quality monitoring procedures are discussed in this section:

1. Visual;
2. Temperature;
3. Algae;
4. Aquatic plants;
5. Water clarity / turbidity;
6. Dissolved oxygen;
7. Sampling for chemical analysis;
8. Sampling for bacteriological analysis; and
9. pH.

Each of the monitoring procedures are described on a separate page, so that you can add information to a monitoring procedure or you can replace a monitoring procedure are newer information becomes available.

2.1 VISUAL ASSESSMENT

Visual monitoring provides a qualitative description of the physical characteristics of the reservoir and banks. Depending on which parameter is being assessed, it may be necessary to collect a sample of water in a clear glass bottle for assessment purposes. The following parameters can be assessed:

- ***Colour:*** Is the water colourless or does it have a colour (e.g. milky, turbid, light brown, dark brown, light blue or green)? Colour in water can be due to pollutants or suspended matter. Look for dramatic changes in water colour when assessing this parameter.
- ***Oil sheen:*** An oil sheen is present if a film of iridescent colour is noted on the surface of the sample. Look for a rainbow effect that can appear to be floating on the surface of the water.
- ***Odour:*** Note whether any odours are present and what they smell like. For example, rotten eggs (hydrogen sulphide), a sour smell, sewage, chlorine, petrol fumes, freshly mown grass or dead fish.
- ***Foam:*** Gently shake the sample and observe any foaming. Foam in the sample is most likely caused by surfactants and may resemble dish-washing soapsuds.
- ***Reservoir bank:*** Is there natural riparian vegetation or is it degraded? Are the banks collapsed and eroded? Is there garbage next to, or within, the reservoir?
- ***Biological characteristics:*** Is there wildlife, such as birds, amphibian, reptiles or mammals, in or around the reservoir? Are there fish in the reservoir? Are there any aquatic plants? Are these aquatic plants attached, limited to the bank, covering the entire surface? Is there any algae in the reservoir? What does the algae look like (e.g. filamentous algae or mats of algae floating on the water's surface)?

2.2 SAMPLING PROCEDURES

2.2.1 General preparation and sampling procedures

Reused sample containers and glassware must be cleaned and rinsed before the first sampling run and after each run. Unless specified in the sampling procedure, the following General Preparation for Sampling Containers can be used.

General Preparation of Sampling Containers
The following method should be used when preparing all sample containers and glassware for chemical analysis, monitoring of turbidity, pH and dissolved oxygen. Wear latex gloves!

1. Wash each sample bottle or piece of glassware with a brush and phosphate-free detergent.
2. Rinse three times with cold tap water.
3. Rinse three times with distilled or deionised water.

2.2.2 Collecting a surface grab sample

This method is based on the one used by DWA using a bucket to collect the sample. It should not be used when an obvious surface scum is present.

Apparatus

- A clean plastic bucket. If the sample is to be taken from the bank, then a 3 m rope should be attached.
- A clean plastic sample bottle with screw cap. Ensure the bottle is clean and clearly marked (site number, date and time and the name of the sampler).

1	• Throw the bucket out into the water. • Using the rope, haul in across the surface filling the bucket with water. • Rinse and discard contents. Repeat this step.	 Rinse twice
2	Again throw the bucket out into the water and haul in collecting the sample.	 Take sample
3	Fill the sample bottle. Ensure the correctly marked sample bottle is used.	 Fill sample bottle
4	Pour out a small amount (about 2 cm from the top of bottle). This allows the sample to be properly mixed before analysis.	 Decant 2 cm
5	Screw cap tightly into place.	 Close tightly

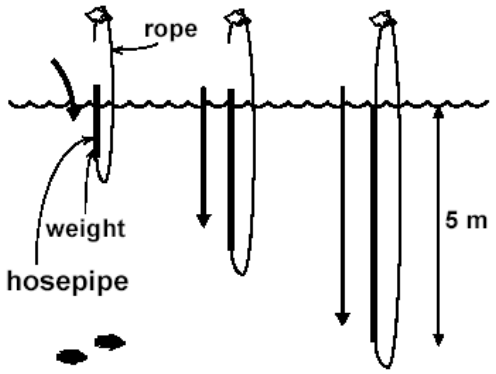
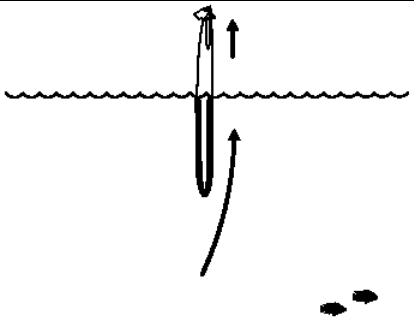
2.2.3 Collecting an integrated sample (5 m hosepipe sample)

An integrated sample provides an indication of the overall population of algae present in the water column. Sampling using this method is only appropriate when the depth at the sampling site is greater than 5 m.

Apparatus:

- A 5 m standard hosepipe (e.g. 15 mm inner diameter), tied at both ends with a single piece of rope. If the sample is being taken from a boat, the rope can be 6 to 7 m long. If the sample will be taken from a point high above the water (like the reservoir wall) then the length of the rope must be at least the height of the wall above the water level plus 5 m. One end of the hosepipe is weighted. Store the hosepipe hanging or lying straight.
- A clean plastic sample bottle with screw cap. Ensure the bottle is clean and clearly marked. If DWA is to perform the analyses, then use the sample bottles supplied by DWA. Ensure that the sampling containers (algal identification, chlorophyll *a*, chemical sampling bottles and the bucket) are clean and clearly marked with the masking tape.

Sample collection procedure:

1	Slowly lower the weighted end of the hosepipe into the water so that it descends straight down (i.e. vertically) until the unweighted end is just below the surface. Ensure that the hosepipe is hanging in a vertical position. If sample is being taken from a boat, ensure that the boat is anchored.	 Lower vertically
2	Using the attached rope, pull up the weighted end (forming a U-shape) until that end reaches the surface. Raise the hosepipe out of the water, maintaining the U-shape.	 Raise in U-shape
3	Fill the sample bottle with water from the hosepipe. Repeat steps 1 and 2 until sufficient sample has been collected (approximately 8 litres). Ensure the correctly marked sample bottle is used.	 Fill sample bottle
4	Pour out a small amount (about 2 cm from the top of bottle). This allows the sample to be properly mixed before analysis.	 Decant 2 cm

Sample preservation procedure (if DWA is the analysing laboratory):

5	• Preserve the sample with mercury chloride ampoules. • Take one ampoule and gently tap the bottom end to ensure that all the liquid is in the wide part of the ampoule. • Hold the ampoule in one hand – use the thumb and index finger of the other hand to snap the neck off. • Place both parts in the sample.	
6	• Screw top tightly into place. • Shake well. • Do not open bottle again.	 Close tightly

MERCURY CHLORIDE – IMPORTANT WARNING AND PRECAUTIONARY MEASURES

- The liquid in the ampoule (mercury chloride) is highly toxic and must be handled with care. Each ampoule contains 6 mg Mercury (II).
- Neither broken nor unbroken ampoules or the contents thereof must under any circumstances be exposed to heat or fire.
- If there was skin contact with the contents of the ampoules, wash the area of contact thoroughly with soap. If an ampoule accidentally breaks, wash the surrounding area with lots of water.
- If the liquid is accidentally swallowed a doctor should be contacted immediately. **FIRST AID:** Drink a mixture of one raw egg white and a glass of milk.
- To minimize the risk of contamination of the water sample the ampoules should be kept clean. Handle it as little as possible and avoid contact with soil, soap etc.
- Keep the ampoules in the containers provided and keep out of reach of other people especially children.
- Old sampling bottles cannot be used for anything else other than water samples as the preservative impregnates the plastic.

2.3 WATER TEMPERATURE

How does temperature affect reservoir characteristics?

No two reservoirs are exactly alike. They may differ in size, depth, number and size of inflowing and out flowing streams and shoreline configuration. Each of these physical factors in turn affects the reservoir character.

In deep reservoirs, water near the surface may be very different physically, chemically and biologically from water near the bottom. The top portion of the reservoir is mixed by the wind and warmed by the sun. The availability of light and warmer temperatures in the top portion creates a preferred habitat for photosynthesising organisms. This top layer is referred to as the "epilimnion[2]". The "metalimnion[3]" is the middle layer of water that marks the transition between the top and bottom layers, where temperature changes rapidly with depth. The bottom portion of a deep reservoir received little of no light. The water is colder, is not mixed by wind and decomposition is the main physical, biological and chemical activity. The bottom layer of water is referred to as the "hypolimnion[4]". This difference in temperatures between the two layers is known as "thermal stratification". When the surface water cools in autumn to the same temperature as the hypolimnion, the stratification is lost and the layers mix. This mixing of the layers is known as "autumn turnover". The mixing of the reservoirs associated with the turnover often corresponds with a large increase n turbidity.

A shallow reservoir is more likely to be homogenous – the same from top to bottom. The water is well mixed by wind and physical characteristics, such as temperature and oxygen vary little with depth. Photosynthesis and growth thus occur throughout the water column as sunlight reaches all the way to the bottom of the reservoir.

Sampling and equipment considerations:

As mentioned above, temperature in deep reservoirs varies horizontally. Recording the change in temperature with depth is best undertaken with a temperature probe attached to a long lead (30 m). The probe is lowered, from the surface of the reservoir, at fixed intervals (normally between 50 cm and 1 m intervals) and the temperature is recorded at each depth. Recordings stop when the bottom is reached or the maximum length of the lead.

Some modern multi-probe instruments can record depth, temperature and dissolved oxygen continuously as the probe is lowered, giving a more accurate portrayal of the changes with depth.

[2] *Epilimnion*: The upper layer of water in a reservoir that is comprised of uniformly warm water that may be well mixed.
[3] *Metalimnion*: The middle layer of water that marks the transition between the top and bottom layers, where temperature changes rapidly with depth.
[4] *Hypolimnion*: The bottom layer of water in a reservoir, which comprises of uniformly cold, stagnant and undisturbed water.

2.4 ALGAE

What are algae and why are they important?

Algae is a collective term referring to a wide range of oxygen producing, photosynthetic organisms that are usually present in surface waters. Algae have no true roots, stems or leaves. Algae range in size from microscopically small single cell to larger mass aggregates of cells (colonies) or in strands (filaments).

The algae are an important living component of lakes. Some of the essential functions which algae provide include:

- Converting inorganic material to organic matter through photosynthesis;
- Oxygenate the water, through the process of photosynthesis;
- Serve as the essential base of the food chain; and
- Affect the amount of light that penetrates into the water column.

Like most plants, algae require light, a supply of inorganic nutrients and specific temperature ranges to grow and reproduce. Of these factors, it is usually the supply of nutrients that will determine the amount of algal growth in a reservoir. In most reservoirs, increasing the supply of nutrients (especially phosphorus) in the water will usually result in a larger algal population, which can become a nuisance and interfere with the desirable uses of the reservoir. This can be a natural phenomenon, but it is often the result of accelerated cultural eutrophication[5] caused by human activities.

What are the factors that affect algal growth?

There are a number of environmental factors that influence algal growth, including:

- The amount of light that penetrates the water (determined by the intensity of sunlight, the turbidity of the water and water colour);
- The availability of nutrients for algal uptake (determined both by source and removal mechanisms);
- Water temperature (regulated by climate, altitude, etc);
- The physical removal of algae by sinking or flushing through an outflow;
- Grazing on the algal population by microscopic animals, fish, and other organisms;
- Parasitism by bacteria, fungi, and other micro-organisms; and
- Competition pressure from other aquatic plants for nutrients and sunlight.

It is a combination of these and other environmental factors that determine the type and quantity of algae found in a reservoir. It is important to note, however, that these factors are always in a state of change. This is because a multitude of events, including the change of seasons, development in the Catchment and rainstorms constantly create "new environments" in a reservoir. These environmental changes may or may not present optimal habitats for growth or even survival for any particular species of algae. Consequently, there is usually a succession of different species in a reservoir over the course of a year and from year to year.

Excessive growth of one or more species of algae is termed a “bloom”. Algal blooms, usually occurring in response to an increased supply of nutrients, are often a disturbing symptom of eutrophication. Blooms of algae can give the water an unpleasant taste or odour, reduce clarity and colour the reservoir a vivid green, brown, yellow, or even red, depending on the

[5] *Cultural eutrophication*: The process of nutrient (especially phosphorus) enrichment, usually resulting from human activities (such as the discharge of effluent, excessive use of fertilizers or detergents).

species. Filamentous and colonial algae are especially troublesome because they can mass together to form scums or mats on the reservoir surface. These mats can drift and clog water intakes, foul beaches, and ruin many recreational opportunities.

Sampling and equipment considerations:

The measurement of algae in reservoirs is usually by means of the chlorophyll *a* concentration in the water. Chlorophyll *a* is the green pigment in plants that allows them to photosynthesis. By measuring chlorophyll *a*, you are indirectly measuring the amount of photosynthesising plants (i.e. algae) found in the sample.

The chlorophyll *a* concentration cannot be considered to be a precise measurement of algae density, because the amount of chlorophyll *a* found in algal cells varies among algal species, depth of the water sample and availability of sunlight. Healthy algal cells constantly try to maintain chlorophyll concentrations at a level for maximum photosynthetic efficiency. For this reason, chlorophyll concentrations usually decrease during high light conditions and increase during the night or low light conditions. Similarly, algal cells that are sinking down into the water column (away from the sun) may also produce more chlorophyll to compensate for lower light levels found at greater depths. Changing seasons also create higher or lower light conditions according to the position of the sun, which in turn, affects chlorophyll production.

Despite these drawbacks, the ease of sampling and relatively low cost of the chlorophyll *a* analysis makes chlorophyll *a* an attractive parameter for characterising the algal density in reservoirs. Chlorophyll *a* is analyzed in a laboratory from a sample collected by a volunteer. A 1 litre water sample is sufficient for chlorophyll a analysis. The sample should not be preserved other than placing the sample in a cooler box on ice.

Visual monitoring *in situ* is also recommended for the early detection of algal blooms. The variables that are monitored include:

1. The presence or absence of algal blooms;
2. The colour of the water; and
3. The total surface area covered by algal blooms.

2.5 AQUATIC PLANTS

Aquatic plants have true roots, stems, and leaves. They, too, are a vital part of the biological community of a reservoir. Unfortunately, like algae, they can overpopulate and interfere with reservoir uses. Excessive water plant growth is generally as a result of eutrophication and/or habitat disturbance. Aquatic plants can be grouped into four categories:

1. Emergent plants are rooted and have stems or leaves that rise well above the water surface. They grow in shallow water or on the immediate shoreline where water lies just below the land surface. They are generally not found in reservoir water over much more than half a metre deep.
2. Rooted floating-leaved plants have leaves that rest on, or slightly above, the water surface. These plants, whose leaves are commonly called lily pads or "bonnets," have long stalks that connect them to the reservoir bottom.
3. Submerged plants grow with all or most of their leaves and stems below the water surface. They may be rooted in the reservoir bottom or free-floating in the water. Most have long, thin, flexible stems that are supported by the water. Most submerged plants flower above the surface.
4. Free-floating plants are found on the reservoir surface. Their root systems hang freely from the rest of the plant and are not connected to the reservoir bottom.

Through photosynthesis, aquatic plants convert inorganic material to organic matter and oxygenate the water. They provide food and cover for aquatic insects, crustaceans, snails, and fish. Aquatic plants are also a food source for many animals. In addition, waterfowl, and other species use aquatic plants for homes and nests.

Aquatic plants are effective in breaking the force of waves and thus reduce shoreline erosion. Emergent plants serve to trap sediments, silt, and organic matter flowing off the catchment. Nutrients are also captured and utilized by aquatic plants, thus preventing them from reaching algae in the open portion of a reservoir.

TASK 1 : PREPARE BEFORE LEAVING FOR THE SAMPLE SITE

Refer to **Part 2** for details on confirming sampling date, time safety considerations, checking supplies and checking weather and directions. In addition to the standard sampling equipment you will need a plastic jar for the sample collection.

TASK 2 : COLLECT THE SAMPLE

Aquatic plants should be carefully taken to ensure that the roots, stems, flowers and seeds are all collected. Plants can be dried between newspaper sheets and forwarded to the National Botanical Institute for identification. Alternatively, submerged plants can be kept in the plastic jar in the fridge for no more than two days before being taken to the National Botanical Institute for identification. In both cases, make sure to include the name of the collector, coordinates if possible or a good description of the locality. Makes notes on where the plant grows and what it looks like.

The DWA publication, Easy Identification of Aquatic Plants (Gerber *et al*., 2004), can be used for field identification of aquatic plants.

2.6 WATER CLARITY

Why is turbidity important?

Turbidity is a measure of water clarity and determined by how much material is suspended in the water. Suspended materials include soil particles (cay slit and sand), algae, plankton, microbes and other substances. Turbidity can also affect the colour of the water. The impacts of high turbidity include:

- High concentrations of particulate matter can cause increased sedimentation and siltation in the reservoir, which can reduce available habitats for fish and other aquatic life.
- Higher turbidity can also increase the water temperature, as suspended particles absorb more heat. This in turn reduces the concentration of dissolved oxygen, because warm water holds less oxygen than cold water.
- Higher turbidity also reduces the amount of light penetrating the water, which reduces the rate of photosynthesis and the production of dissolved oxygen.
- Suspended materials may also clog fish gills, reducing the resistance to disease in fish, lowering growth rates and affecting egg and larval development.
- Suspended solids can also provide attachment places for pollutants, such as metals and bacteria.

What are the sources of suspended materials (i.e. turbidity)

Land use is probably the greatest factor influencing changes in turbidity in rivers. As Catchment areas develop, there is an increase in disturbed areas and a decrease in vegetation, which in turn increases the rate of run-off water. Turbidity is therefore a useful indicator of the effects of run-off from construction, agricultural practices, discharges and other land use practices. Other sources of turbidity include:

- Soil erosion;
- Waste discharge;
- Urban run-off;
- Eroding stream banks;
- Large numbers of bottom feeders (such as carp), which stir up bottom sediments; and
- Excessive algal growth.

Sampling and equipment considerations:

Turbidity of not a direct measure of the amount of suspended solids present, although these two are related, or the rate of sedimentation of a river, it is only a measure of the clarity of the reservoir. Turbidity in a reservoir is measured using a Secchi disk, which provides an indication of the depth of light penetration into the water (i.e. measurements are recorded in meters).

A Secchi disk is simply a weighted circular disk 20 cm in diameter with four alternating black and white sections painted on the surface. The disk is attached to a measured line that is marked in meters and sub-divided by tenths of a meter. The Secchi disk is used to measure how deep a person can see into the water. A description of how to make a Secchi disc is provided in **Appendix 3**.

TASK 1 : PREPARE BEFORE LEAVING FOR THE SAMPLE SITE

Refer to **Part 2** for details on confirming sampling date, time safety considerations, checking supplies and checking weather and directions. In addition to the standard sampling equipment you will need:

- A Secchi disk; and
- A data sheet for recording temperature readings.

TASK 2 : TAKING THE MEASUREMENT USING A SECCHI DISK

1. Check to make sure that the Secchi disk is securing attached to the measuring line. Lean over the shady side of the boat and lower the Secchi disk into the water, keeping your back toward the sun to block the glare.

2. Continue to lower the Secchi disk until it disappears from view. Lower the disk another one third of a meter and then slowly raise the disc until it just reappears.

3. Continue to move the disk up and down until the exact vanishing/reappearing point is found. Attached a clothespin to the line at the point where the line enters the water. This is the point at which the measurement will be read.

4. Slowly pull the disk out of the water and record the measurement based on the location of the clothespin line.

2.7 SAMPLING FOR CHEMICAL ANALYSIS

A water resource contains a host of constituents including:

- Physical soil and clay particles and organic detritus from storm runoff;
- Microorganisms, such as bacteria, viruses and parasites;
- Chemical constituents, which can be subdivided into (a) major inorganic chemical salts (such as sodium, chloride, calcium, sulphate, etc.), (b) minor inorganic chemical salts (such as ammonia, fluoride, phosphate and trace metals such as iron, manganese, copper, etc.) and (c) organic substances such as pesticide residues; and
- Radioactive substances (which usually occur only in minute concentrations under natural conditions).

These constituents, and their respective concentration within the water resource, determine the fitness for a variety of uses (domestic, recreation, agriculture and industry) and for the protection of the health and integrity of aquatic ecosystems.

The chemical composition is a function of the Catchment geology and climate. Human activities, such as the discharge of effluent from mines, wastewater treatment works and industries and run-off from agricultural land, also play a big role in determining the chemical constituents of the water resource. A typical freshwater resource has a balance of three major anions and four major cations.

Sampling and equipment considerations:

Chemical analysis is generally done in a laboratory. Macro Sample is the most common type of sample which is taken and analysed for major inorganic ions. The ions most commonly found in natural water resources and provided in **Table 1.4**. The results provide an indication of the ion balance and the inorganic chemical water quality of a water resource.

TASK 1 : PREPARE BEFORE LEAVING FOR THE SAMPLE SITE

Refer to **Part 2** for details on confirming sampling date, time safety considerations, checking supplies and checking weather and directions. In addition to the standard sampling equipment you will need:

- Labels for sample bottles;
- Clean sample bottles, which have been washed in special phosphate free cleaning agents; and
- The preservative (mercury chloride) which is used to fix the sample if DWA is the analysing laboratory.

TASK 2 : COLLECT SAMPLES

Collecting samples from a boat:

- Collect the sample by lowering the bottle into the water. Perform a “forward scoop” motion if the boat is not moving.
- Never take a sample near the motor of the boat.

Sampling from a dam wall:

- Use a clean bucket to collect water. Make sure not to scrape the bucket against the concrete wall when pulling it up.
- Fill the sample bottles immediately.
- Do not sample at a point where lots of debris have accumulated.

Sampling procedure:

- Leave the lid on the bottle until you are ready to take the sample.
- Collect a little bit of water in the sample bottle and rise the bottle. Pour the water out, away from where the sample will be taken.
- Collect the sample by lowering the bottle into the water. Perform a “forward scoop” motion to collect the sample. Do not fill the bottle right up to the top. Leave space for the preservative.
- Preserve the sample with one ampoule containing mercury chloride. Break the ampoule and the drop both pieces into the sample bottle (If DWA is the analysing laboratory).
- Label the sample bottle.
- Store the sample bottle in a cooler box or dark container.

2.8 DISSOLVED OXYGEN

Why is dissolved oxygen important?

Like all terrestrial animals, fish and other aquatic organisms need oxygen to live. In rivers, oxygen is dissolved in water and is either lost or gained through various processes. Dissolved oxygen (DO) is gained from the atmosphere and from plants as a result of photosynthesis. While DO is lost through respiration by aquatic animals, decomposition and various chemical reactions. A certain minimum amount of DO must be present in the water for aquatic life to be sustained.

Why does the level of DO vary?

Oxygen is produced during photosynthesis and consumed during respiration and decomposition. Because photosynthesis requires light, oxygen is only produced by plants during the daylight hours. On the other hand, decomposition and respiration occur 24 hours a day. During the night, when there is no photosynthesis, the DO concentrations steadily decline. They are lowest just below dawn, when photosynthesis resumes. This difference in the production and consumption of oxygen can account for the large daily variation in DO levels.

DO concentration increases wherever water flows becomes turbulent, such as in a rapid or below a waterfall. Another physical process which also impacts on DO concentrations is the temperature of the water. Cold water can hold more DO than warm water. So during the summer months, when the river is warmer, the DO concentration can be limited by the ability of the water to “soak up” more oxygen (**Table 1.2**, page 16). It is therefore important to note the time of your DO sampling to help judge when in the daily cycle the data was collected.

Pollution tends to cause a decrease in DO levels. This can be caused by the addition of effluent or run-off water with a low concentration of DO or chemical or biological constituents that have a high oxygen demand (i.e. they require large amounts of oxygen for before they can be decomposed). The amount of oxygen consumed by these organisms which decompose the chemical or biological constituents is known as the biochemical oxygen demand (BOD).

Sampling and equipment considerations:

In rivers, the DO levels are most likely to vary horizontally along the course of the waterway. The DO levels in and below rapids and waterfalls, are typically higher than those in pools and slower moving stretches.

Dissolved oxygen is measured primarily either by using some variation of the Winkler method or by using a dissolved oxygen meter and probe. DO is measured either in milligrams per litre (mg/l) or "percent saturation." DO measured in mg/l provides an indication of the amount of oxygen in a liter of water. Percent saturation is the amount of oxygen in a litre of water relative to the total amount of oxygen that the water can hold at that temperature.

DO samples are collected using a special BOD bottle: a glass bottle with a "turtleneck" and a ground glass stopper. You can fill the bottle directly in the stream if the stream is wadable or boatable, or you can use a sampler that is dropped from a bridge or boat into water deep enough to submerse the sampler. Samplers can be made or purchased

The Winkler method involves filling a sample bottle completely with water (no air is left to bias the test). The dissolved oxygen is then "fixed" using a series of reagents that form an acid compound that is titrated. Titration involves the drop-by-drop addition of a reagent that

neutralizes the acid compound and causes a change in the colour of the solution. The point at which the colour changes is the "endpoint" and is equivalent to the amount of oxygen dissolved in the sample. The sample is usually fixed and titrated in the field at the sample site. It is possible, however, to prepare the sample in the field and deliver it to a lab for titration.

A dissolved oxygen meter is an electronic device that converts signals from a probe that is placed in the water into units of DO in milligrams per litre. Most meters and probes also measure temperature. The probe is filled with a salt solution and has a selectively permeable membrane that allows DO to pass from the stream water into the salt solution. The DO that has diffused into the salt solution changes the electric potential of the salt solution and this change is sent by electric cable to the meter, which converts the signal to milligrams per litre.

TASK 1 : PREPARE BEFORE LEAVING FOR THE SAMPLE SITE

Refer to **Part 2** for details on confirming sampling date, time safety considerations, checking supplies and checking weather and directions. In addition to the standard sampling equipment you will need:

If using the Winkler Method:

- Labels for sample bottles;
- Field kit and instructions for DO testing;
- Enough reagents for the number of sites to be tested;
- Kemmerer, Van Doorn or home-made sampler to collect deep-water samples;
- A numbered glass BOD bottle with a glass stopper (one for each site); and
- Data sheets to record DO results.

If using a DO meter and probe:

- DO meter and probe (electrode) (NOTE: Confirm that the meter has been calibrated according to the manufacturer's instructions);
- Operating manual for the meter and probe;
- Extra membranes and electrolyte solution for the probe;
- Extra batteries for the meter;
- Extension pole; and
- Data sheet to recode DO results.

TASK 2 : COLLECT SAMPLES

Winkler Method

Use a BOD bottle to collect the water sample. The most common sizes are 300 milliliters (mL) and 60 mL. Be sure that you are using the correct volume for the titration method that will be used to determine the amount of DO. There is usually a white label area on the bottle, and this may already be numbered. If so, be sure to record that number on the field data sheet. If your bottle is not already numbered, place a label on the bottle (not on the cap because a cap can be inadvertently placed on a different bottle) and use a waterproof marker to write in the site number.

If you are collecting duplicate samples, label the duplicate bottle with the correct code, which should be determined prior to sampling by the lab supplying the bottles. Use the following procedure for collecting a sample for titration by the Winkler method:

1. Carefully wade into the stream. Stand so that you are facing one of the banks.

2. Collect the sample so that you are not standing upstream of the bottle. Remove the cap of the BOD bottle. Slowly lower the bottle into the water, pointing it downstream, until the lower lip of the opening is just submerged. Allow the water to fill the bottle very gradually, avoiding any turbulence (which would add oxygen to the sample). When the water level in the bottle has stabilized (it won't be full because the bottle is tilted), slowly turn the bottle upright and fill it completely. Keep the bottle under water and allow it to overflow for 2 or 3 minutes to ensure that no air bubbles are trapped.

3. Cap the bottle while it is still submerged. Lift it out of the water and look around the "collar" of the bottle just below the bottom of the stopper. If you see an air bubble, pour out the sample and try again.

4. “Fix” the sample immediately following the directions in your kit:

    a. Remove the stopper and add the fixing reagents to the sample.

    b. Immediately insert the stopper so air is not trapped in the bottle and invert several times to mix. This solution is caustic. Rinse your hands if you get any solution on them. An orange-brown flocculent precipitate will form if oxygen is present.

    c. Wait a few minutes until the floc in the solution has settled. Again invert the bottle several times and wait until the floc has settled. This ensures complete reaction of the sample and reagents. The sample is now fixed, and atmospheric oxygen can no longer affect it. If you are taking the sample to the lab for titration, no further action is necessary. You can store the sample in a cooler for up to 8 hours before titrating it in a laboratory. If you are titrating the sample in the field, see **Task 3: Analyze the Samples**.

Using a DO meter and probe

If you are using a dissolved oxygen meter, be sure that it is calibrated immediately prior to use. Check the cable connection between the probe and the meter. Make sure that the probe is filled with electrolyte solution, that the membrane has no wrinkles, and that there are no bubbles trapped on the face of the membrane. You can do a field check of the meter's accuracy by calibrating it in saturated air according to the manufacturer's instructions. Or, you can measure a water sample that is saturated with oxygen, as follows. (NOTE: You can also use this procedure for testing the accuracy of the Winkler method.)

1. Fill a 1-liter beaker or bucket of tap water. Mark the bottle number as "tap" on the lab sheet.

2. Pour this water back and forth into another beaker 10 times to saturate the water with oxygen.

3. Use the meter to measure the water temperature and record it in the water temperature column on the field data sheet.

4. Find the water temperature of your "tap" sample in **Table 1.2**. Use the meter to compare the dissolved oxygen concentration of your sample with the maximum concentration at that temperature in the table. Your sample should be within 0.5 mg/l. If it is not, repeat the check and if there is still an error, check the meter's batteries and follow the troubleshooting procedures in the manufacturer's manual.

Once the meter is turned on, allow 15 minute equilibration before calibrating. After calibration, do not turn the meter off until the sample is analyzed. Once you have verified that the meter is working properly, you are ready to measure the DO levels at the sampling site. You might need an extension pole (this can be as simple as a piece of wood) to get the probe to the proper sampling point. Simply secure the probe to the end of the extension pole. To use the probe, proceed as follows:

1. Place the probe in the stream below the surface.

2. Set the meter to measure temperature, and allow the temperature reading to stabilize. Record the temperature on the field data sheet.

3. Switch the meter to read dissolved oxygen.

4. Record the dissolved oxygen level on the field data sheet.

TASK 3 : ANALYSING THE SAMPLES

Three types of titration apparatus can be used with the Winkler method: droppers, digital titrators, and burets. The dropper and digital titrator are suited for field use. The buret is more conveniently used in the laboratory. Volunteer programs are most likely to use the dropper or digital titrator. For titration with a dropper or syringe, which is relatively simple, follow the manufacturer's instructions.

The following procedure is for using a digital titrator to determine the quantity of dissolved oxygen in a fixed sample:

1. Select a sample volume and sodium thiosulfate titration cartridge for the digital titrator corresponding to the expected dissolved oxygen concentration according to **Table 1.2**. In most cases, you will use the 0.2 N cartridge and the 100 ml sample volume.

2. Insert a clean delivery tube into the titration cartridge.

3. Attach the cartridge to the titrator body.

4. Hold the titrator with the cartridge tip up. Turn the delivery knob to eject air and a few drops of titrant. Reset the counter to 0 and wipe the tip.

5. Use a graduated cylinder to measure the sample volume (from the "fixed" sample in the 300 ml BOD bottle) according to **Table 1.3**.

6. Transfer the sample into a 250 ml Erlenmeyer flask, and place the flask on a magnetic stirrer with a stir bar. If you are in the field, you can manually swirl the flask to mix.

7. Place the delivery tube tip into the solution and turn the stirrer on to stir the sample while you're turning the delivery knob.

8. Titrate to a pale yellow color.

9. Add two dropperfuls of starch indicator solution and swirl to mix. A strong blue color will develop.

10. Continue to titrate until the sample is clear. Record the number of digits required. (The colour might reappear after standing a few minutes, but this is not a cause for concern. The "first" disappearance of the blue colour is considered the endpoint.)

11. Calculate mg/l of DO = digits required X digit multiplier (from **Table 1.2**).

12. Record the results in the appropriate column of the data sheet.

Some water quality standards are expressed in terms of percent saturation. To calculate percent saturation of the sample:

1. Find the temperature of your water sample as measured in the field.

2. Find the maximum concentration of your sample at that temperature as given in **Table 1.2**.

3. Calculate the percent saturation, by dividing your actual dissolved oxygen by the maximum concentration at the sample temperature.

4. Record the percent saturation in the appropriate column on the data sheet.

2.9 SAMPLING FOR BACTERIOLOGICAL ANALYSIS

Why are bacteria important?

In order to determine where the river has been contaminated by sewage, members of two bacteria groups that are commonly found in human and animal faeces, coliforms and fecal streptococci, are used as indicators. Although these bacteria are generally not harmful themselves, they indicate the possible presence of pathogenic (disease-causing) bacteria, viruses and protozoa that also live in human and animal digestive systems. Therefore, their presence in streams suggests that pathogenic micro-organisms might also be present and that swimming and eating shellfish might be a health risk.

Since it is difficult, time-consuming and expensive to test directly for the presence of a large variety of pathogens, water is usually tested for indicator organisms instead. Sources of faecal contamination to surface waters include wastewater treatment plants, on-site septic systems, domestic and wild animal manure and storm runoff. In addition to the possible health risk associated with the presence of elevated levels of faecal bacteria, they can also cause cloudy water, unpleasant odours and an increased oxygen demand.

The most commonly tested faecal bacteria indicators include total coliforms, faecal coliforms, *Escherichia coli* and enterococci. Each indicator bacteria is used for specific monitoring purposes, which are described below.

- *Total coliform bacteria*: Primarily used as a practical indicator of the general hygienic quality of water. Mainly used in routine monitoring of drinking water supplies.
- *Faecal coliform bacteria*: Primarily used as a practical indicator of faecal pollution, more specific for faecal pollution than total coliforms. Mainly used for assessment of faecal pollution of wastewater, raw water supplies and natural water environments used for recreational purposes.
- *Escherichia coli*: Highly specific indicator of faecal pollution which originates from humans and warm-blooded animals.
- *Enterococci* (*faecal streptococci*): Relatively specific indicators of faecal pollution which tend to survive longer in water environments than coliform bacteria.

Sampling and equipment considerations:

Bacteria can be difficult to sample and analyse, for many reasons including:

- Natural variation of bacteria levels;
- Weather conditions, bacteria levels are strongly correlated with rainfall, and thus comparing wet and dry weather bacteria data can be a problem;
- Many analytical methods have a low level of precision yet can be quite complex; and
- Absolutely sterile conditions are required to collect and handle samples.

The primary equipment decision to make when sampling for bacteria is what type and size of sample container you will use. Once you have made that decision, the same, straightforward collection procedure is used regardless of the type of bacteria being monitored. It is critical when monitoring bacteria that all containers and surfaces with which the sample will come into contact be sterile. Containers made of either some form of plastic or Pyrex glass are acceptable. However, if the containers are to be reused, they must be sterilized using heat and pressure. Plastic containers, made from either high-density polyethylene or polypropylene, might be preferable to glass from a practical standpoint because they will better withstand breakage. The size of the container will depend on the sample amount

needed for the bacteria analysis method you choose and the amount needed for other analyses. There are two basic methods for analysing water samples for bacteria:

1. The membrane filtration method involves filtering several different-sized portions of the sample using filters with a standard diameter and pore size, placing each filter on a selective nutrient medium in a petri plate, incubating the plates at a specified temperature for a specified time period, and then counting the colonies that have grown on the filter. This method varies for different bacteria types (variations might include, for example, the nutrient medium type, the number and types of incubations, etc.).

2. The multiple-tube fermentation method involves adding specified quantities of the sample to tubes containing a nutrient broth, incubating the tubes at a specified temperature for a specified time period, and then looking for the development of gas and/or turbidity that the bacteria produce. The presence or absence of gas in each tube is used to calculate an index known as the Most Probable Number (MPN).

Given the complexity of the analysis procedures and the equipment required, field analysis of bacteria is not recommended.

TASK 1 : PREPARE BEFORE LEAVING FOR THE SAMPLE SITE

Refer to **Part 2** for details on confirming sampling date, time safety considerations, checking supplies and checking weather and directions. In addition to the standard sampling equipment you will need:

1. Labels for sample bottles; and
2. Sterilised sample bottles.

TASK 2 : COLLECT SAMPLES

Samples should be collected using the standard procedure described in **Part 2**. Note that sample bottles must be sterilised before they are used. Be careful not to touch the inside of the bottle or the lid.

Remember to wash your hands thoroughly after collecting samples suspected of containing faecal contamination. Also be careful not to touch your eyes, ear, nose or mouth until your have washed your hands.

Samples for bacteria must be analysed within six hours of collection. Keep the samples on ice and take them to the laboratory as soon as possible.

2.10 PH

What is pH and why is it important?

The pH of a sample is a terms used to indicate the alkalinity or acidity of a substance ranked on a scale from 1.0 to 14.0. A pH of 7 is considered to be neutral. Substances with pH less than 7 are acidic, while substances with pH greater than 7 are basic. The pH of most natural waters ranges between 6.5 and 8.5.

The pH of water determines the solubility (amount that can be dissolved in the water) and biological availability (amount that can be utilised by aquatic life) of chemical constituents, such as nutrients (e.g. phosphorus, nitrogen and carbon) and heavy metals (e.g. lead, cadmium and copper). For example, pH can determine how much and in what form phosphorus is most abundant in the water. pH can also allow toxic elements and compounds to become mobile and "available" for uptake by aquatic animals and plants. pH also affects biological processes in water. For example, different organisms flourish within different ranges of pH. The largest variety of aquatic organisms prefers a pH range between 6.5 and 8.0. pH outside this range reduces the diversity in the river because it stresses the organisms and can reduce reproduction.

The pH scale measures the logarithmic concentration of hydrogen (H^+) and hydroxide (OH^-) ions, which make up water ($H^+ + OH^- = H_2O$). When both types of ions are in equal concentration, the pH is 7.0 or neutral. Below 7.0, the water is acidic (there are more hydrogen ions than hydroxide ions). When the pH is above 7.0, the water is alkaline, or basic (there are more hydroxide ions than hydrogen ions). Since the scale is logarithmic, a drop in the pH by 1.0 unit is equivalent to a 10-fold increase in acidity. So, a water sample with a pH of 5.0 is 10 times as acidic as one with a pH of 6.0, and pH 4.0 is 100 times as acidic as pH 6.0.

Why does the pH vary?

The geology of the catchment and the original source of the water determines the initial pH of the water. The greatest natural cause for change in pH is the seasonal and daily variation in photosynthesis. Photosynthesis uses up hydrogen molecules, which causes the concentration of H+ ions to decrease and therefore the pH increases. Respiration and decomposition processes lower the pH. For this reason pH is higher during daylight hours and during the growing seasons when photosynthesis is at its peak. Although pH may constantly change, the amount of change is remains within a narrow pH range. Natural waters also have the ability to prevent major changes in pH. Small or localised changes in pH are quickly modified by various chemical reactions so little or no change may be measures. The ability to resist change in pH is called "buffering capacity".

Industrial activities generally cause acidification rather than alkalinisation of rivers. Acidification is normally the result of three different types of pollution, namely:

- Low pH point-source effluents from industries, such as pulp and paper and tanning and leather industries;
- Mine drainage, which is nearly always acid, leading to the pH of receiving streams dropping to below two; and
- Acid precipitation resulting largely from atmospheric pollution caused by the burning of coal (and subsequent production of sulphur dioxide (SO_2)) and the exhausts of combustion engines (nitrogen oxides (NO_x)). Both sulphur oxides (SO_x) and nitrogen oxides (NO_x) form strong mineral acids when dissolved in water. When acid rain falls on a catchment, the strong acids leach calcium and magnesium from the soil and also interfere with nutrient availability.

Elevated pH values can be caused by increased biological activity in eutrophic systems. The pH values may fluctuate widely from below six above ten over a 24-hour period as a result of changing rates of photosynthesis and respiration.

Sampling and equipment considerations:

pH can be analyzed in the field or in the lab. If it is analyzed in the lab, you must measure the pH within two hours of the sample collection. This is because the pH will change due to the carbon dioxide from the air dissolving in the water, which will bring the pH toward seven. Alternatively, pH measurements can be taken in the field using pH meters, colour comparators or pH "pocket pals".

pH Meters

A pH meter measures the electric potential (millivolts) across an electrode when immersed in water. This electric potential is a function of the hydrogen ion activity in the sample. Therefore, pH meters can display results in either millivolts (mV) or pH units. A pH meter consists of a potentiometer, which measures electric current; a glass electrode, which senses the electric potential where it meets the water sample; a reference electrode, which provides a constant electric potential; and a temperature compensating device, which adjusts the readings according to the temperature of the sample (since pH varies with temperature). The reference and glass electrodes are frequently combined into a single probe called a combination electrode. There is a wide variety of meters, but the most important part of the pH meter is the electrode.

pH "Pocket Pals" and Colour Comparators

pH "pocket pals" are electronic hand-held "pens" that are dipped in the water and provide a digital readout of the pH. They can be calibrated to one pH buffer. Colour comparators involve adding a reagent to the sample that colours the sample water. The intensity of the colour is proportional to the pH of the sample. This colour is then matched against a standard colour chart. The colour chart equates particular colours to associated pH values. The pH can be determined by matching the colours from the chart to the colour of the sample.

TASK 1 : PREPARE SAMPLE CONTAINERS

Sample containers (and all glassware used in this procedure) must be cleaned and rinsed before the first run and after each sampling run by following the procedure described in **Part 2**.

TASK 2 : PREPARE BEFORE LEAVING FOR THE SAMPLING SITE

Refer to **Part 2** for details on confirming sampling date, time safety considerations, checking supplies and checking weather and directions.

In addition to the standard sampling equipment you will need:

- pH meter with combination temperature and reference electrode, or pH "pocket pal" or colour comparator;
- Wash bottle with deionised water to rinse pH meter electrode (if appropriate); and
- Data sheet for pH to record results.

Before you leave for the sampling site, be sure to calibrate the pH meter or "pocket pal." The pH meter and "pocket pal" should be calibrated prior to sample analysis and after every 25 samples according to the instructions that come with them.

TASK 3 : COLLECT THE SAMPLE

Samples are collected using the standard sampling process described in **Part 2**. Which sampling procedure should be used?

TASK 4 : MEASURE pH

The procedure for measuring pH is the same whether it is conducted in the field or lab. If you are using a "pocket pal" or colour comparator, follow the manufacturer's instructions. Use the following steps to determine the pH of your sample if you are using a meter.

1. Rinse the electrode well with deionised water.

2. Place the pH meter or electrode into the sample. Depress the dispenser button once to dispense electrolyte.

3. Read and record the temperature and pH in the appropriate column on the data sheet. Rinse the electrode well with deionised water.

4. Measure the pH of the 4.01 and 7.0 buffers periodically to ensure that the meter is not drifting off calibration. If it has drifted, recalibrate it.

3. FISH HEALTH ASSESSMENT

The status of a fish population provides a good indication not only of the status of the ecosystem health, but also the potential status of human health, especially amongst communities which have access to rivers and reservoirs. The consumption of fish is generally beneficial to humans as they provide an excellent source of protein and vitamins. Other health benefits include a decrease in cardiovascular disease, reduced risk of colon and breast cancer and a reduction in high blood pressure. The pollution of freshwater aquatic systems by point source discharges (such as the discharge of effluent from wastewater treatment works and industrial effluent) and diffuse surface run-off (agriculture, mining and urban) results in the bioaccumulation of chemicals by freshwater fish. If these levels of contamination in the fish become too high, then human consumption of the fish can result in a human health risk.

3.1 FISH HEALTH OBSERVATION

Fish health observation is a rapid visual assessment of the status of the fish population. The following variables should be assessed:

- Condition of skin;
- Condition of fins;
- Condition of eyes;
- Condition of opercula (gill covers);
- Condition of gills; and
- The number of ectoparasites.

Table 2.1 provides an overview of the conditions which should be noted when conducting a fish health observation.

Table 2.1: Fish health assessment index variables

Variable	Variable condition
Skin	• Normal, no aberrations • Mild skin aberrations • Moderate skin aberrations • Sever skin aberrations
Fins	• No active erosion or previous erosion healed over • Mild active erosion with no bleeding • Sever active erosion with haemorrhage / secondary infection
Eyes	• Normal – no aberrations evident (good “clear” eyes) • Exopthalmia – swollen, protruding eye • Hemorrhagic – bleeding in the eye • Blind • Missing – eye missing from the fish • Other – any manifestations which could not “fit” the above
Opercules	• No shortening – normal • Mild shortening • Sever shortening
Gills	• Normal – no apparent aberration in gills • Frayed – erosion of tips of gill lamellae “ragged” look • Clubbed – swelling of the tips of the gill lamellae • Marginate – gill with a light discoloured margin along the distal end or tips of the lamellae of filament • Pale – gills are definitely very light in colour • Other – any observation which does not fit the above

Ectoparasites	• No parasites observed • 1 – 10 parasites • 11 – 20 parasites • > 20 parasites

3.2 FISH KILLS

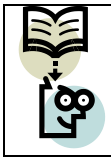
Fish die as a result of a wide variety of natural and unnatural causes. A few dead fish floating on the surface of a pond or dam is not necessarily cause for alarm. However, when large numbers of fish of all sizes are found dead and dying over a long period of time, it is necessary to investigate and determine the cause. Natural causes of fish kills include:

- Oxygen depletion;
- Oxygen super-saturation;
- Toxic algal blooms;
- Autumn turnover;
- Sudden temperature changes;
- Salinity changes;
- Bacterial infections;
- Viruses; and
- Parasites.

Man induced factors include the discharge of pesticides / fertilisers / other chemicals into the water which can kill fish or lower available dissolved oxygen.

Report a fish kill to the DWA: Regional Office as soon as possible. The following information should be supplied:

- An estimate of the number of fish dead/dying;
- Time when kill was first noticed;
- What other organisms were affected;
- The species of fish affected;
- The size of the fish affected;
- The physical extent of the area affected;
- Possible causes; and
- What possible sources of pollution are in the area.



Additional reading material:

- DWAF: Resource Quality Services. 2004. Water Resource Quality Monitoring. Volume 5: Fish Kill Investigation.

4. EXAMPLES OF OTHER VOLUNTEER MONITORING PROGRAMMES

4.1 UNITED STATES VOLUNTEER MONITORING

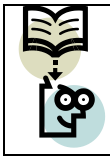
Across the country, trained citizens are monitoring the conditions of their local streams, lakes, estuaries and wetlands. The US Environmental Protection Agency (EPA) has developed a website for volunteer monitoring, which provides information and encouragement to all citizens to learn about their water resources. Volunteer monitors build awareness of pollution problems, become trained in pollution prevention, help clean up problem sites, provide data for water resources that may otherwise be unassessed and increase the amount of water quality information available to decision makers at all levels of government.

The EPA volunteer website provides useful information on water quality monitoring procedures and fact sheets on starting out in volunteer monitoring.

4.2 WATERWATCH AUSTRALIA

Waterwatch is a national community-based water quality monitoring programme that encourages all Australians to become active in the protection of their waterways. The Waterwatch program was established by the Australian Government during 1993. There are now nearly 3 000 Waterwatch groups monitoring water quality at over 7 000 sites throughout 200 catchments. Waterwatch groups conduct biological and habitat assessments plus physical and chemical water tests.

The Waterwatch website provides information on starting a monitoring programme, safety guidelines, biological parameters (macro-invertebrate monitoring and undertaking habitat surveys) and physical and chemical parameters that should be assessed.



Additional reading material:

- US EPA volunteer website: http://www.epa.gov/volunteer/
- Australian Waterwatch website: http://www.waterwatch.org.au

PART 4 : STREAM FLOW CONDITIONS INVENTORY

This chapter describes a simple method for estimating the flow in a stream. Stream flow estimates are useful for calculating the load of a particular substance in that stream.

1. FLOW GAUGING / ESTIMATION

The water generally comes from surface run-off and from water that has passed through the soil and out into the waterway. The amount of any particular substance carried in the water is known as the load. The faster and bigger the flow of the water, the stronger the current, and the heavier the load it can carry.

When there is little water in the waterway (low flow) most of the water entering the stream will be from underground seepage, and the flow rate is slow. Sediment settles quickly to the bottom, sections of the stream will become semi-stagnant resulting in low dissolved oxygen concentrations, algal growth will increase if there is adequate light, leading to algal blooms, and salinity and water temperature may increase to values that affect the biota in the waterway.

Moderate flows ensure good mixing of oxygen with water, and dilution and flushing of contaminants.

After heavy rainfall the water level rises or floods (high flow) because run-off rushes into the waterway increasing turbidity and the load of contaminants. During flooding, the concentrations of oxygen, turbidity, pH, salinity and nutrients can be expected to fluctuate.

For the purposes of measurement, flow is the velocity of water multiplied by the cross-sectional area of the stream. These two quantities must be measured as accurately as possible to avoid compounding errors when calculating flow.

What factors affect flow?
Flow is modified by conditions along and around the waterway, such as:

- structures, such as dams and weirs, in the waterway;
- removal (diversion) of water for use in irrigation, industry and households;
- rainfall, snow melt, and water releases from dams and power stations;
- entry of groundwater;
- evaporation; and
- the leakiness of the river bed and banks.

The size of a waterway and its flow rate affect its water quality. For example, discharges containing contaminants will have less effect on large swiftly flowing rivers than on small slow streams. This is one reason for measuring flow - to work out the load of contaminants and sediment the waterway is carrying.

Because velocity and flow have a significant effect on water quality, it is important that you record them at the time of sampling. It is particularly valuable to know if flows are at low, moderate or high level and if the level is rising or falling. This is because the concentrations of

nutrients, turbidity and contaminants tend to be higher when the stream level is rising than when it is falling.

Methods and equipment considerations:

There are many ways to measures flow, the two simplest ways are described below:

1. A simple method is to see how fast a floating object travels downstream over a chosen distance. This is called the float method.
2. Flow data can be obtained from the Regional DWA Office, if your site is near a gauging station. The Regional DWA office measures and keeps records of flow on a regular basis at gauging stations spaced out along all main waterways.

Float method for determining water velocity:

The float method is easy to understand and something most of us have done as children. You simply float an object on the water and measure the time it takes to travel a set distance. The equipment you will need for this method includes:

- tennis ball, apple or orange
- net to catch the ball or fruit
- 10 metre tape or rope
- stopwatch

Procedure

1. Mark out a 10-metre length of the river, upstream of your sampling site. Choose a section of the river that is relatively straight and free of vegetation or obstacles. Avoid areas with a culvert or bridge because those structures will modify the true flow. If the flow is very slow, mark out a shorter distance.

2. Position a person at each end of the 10-metre section.

3. Place the ball on the surface near the middle of the river at least two metres upstream of the end of the tape so it has time to come up to water speed.

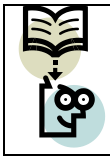
4. When the ball is in line with the beginning of the tape, start the stopwatch.

5. Stop the watch when the ball gets to the end of the 10-metre section.

6. Repeat the procedure at least three times at this site and average the results.

7. To calculate the water velocity, divide the distance travelled in metres by the time taken in seconds. Then multiply by a correction factor of 0.9 to compensate for the variability in velocity with depth and across the channel, i.e. water will flow more slowly at the edges than in the middle, and more slowly near the bottom than near the surface.

Stream velocity = (distance travelled x correction factor) ÷ average time taken



Additional reading material:

- United States Environmental Protection Agency. Volunteer Stream Monitoring: A Methods Manual. http://www.epa.gov/owow/monitoring/volunteer/stream/

PART 5 : REPORTING POLLUTION INCIDENTS

This chapter describes who should be contacted to report a pollution incident and what information should be provided. Pollution incidents that occur within the boundaries of a local municipality should be reported to that municipality. Pollution incidents that fall outside the boundaries of a local municipality should be reported to the DWA.

If you become aware of a pollution incident, it is important to know which authority to report it to. Contacting the right authority and providing enough information about the incident will increase the opportunity for prompt and effective action.

1. WHAT IS A POLLUTION INCIDENT

In terms of the National Water Act, 1998 (Act No. 36 of 1998), "waste" *includes any solid material or material that is suspended, dissolved or transported in water (including sediment) and which is spilled or deposited on land or into a water resource in such a volume, composition or manner as to cause, or to be reasonably likely to cause, the water resource to be polluted*.

Solid waste – Solid waste can be any material that is dumped in a water course that can be harmful to the aquatic environment or human health or unreasonably interfere with the enjoyment of water bodies such as streams, rivers, dams or wetlands. Examples of solid waste include litter, discarded building waste, domestic or industrial waste material, etc.

Liquid waste – Liquid waste refers to liquids being discharged, either by accident or deliberately, that is harmful to the receiving water body, to human health or causes a public nuisance. Examples of liquid waste include leaking or surcharging sewers, discarded oil or paint, domestic waste water, untreated sewage, untreated industrial waste water, etc.

Transport accidents – pollution can also result from road or rail accidents involving trucks or trains transporting fuel or chemicals.

Pollution incidents can be once-off events (such as an accident), intermittent discharges (such as polluters discharging waste at night or over weekends), or continuous discharges of waste water into streams and rivers (such as leaking sewers or seepage from wastewater dams).

2. WHO TO CONTACT

Pollution within the boundaries of a local authority - If a pollution incident falls within the boundaries of a local municipality or city council, you need to contact the department that deals with stormwater and/or sewerage. Many local authorities and city councils have call centres that ensures that incident reports are routed to the correct municipal department.

Pollution outside the boundaries of a local authority - If the pollution incident falls outside of the boundaries of a local authority, you need to contact the DWA. The Department has regional offices in many parts of the country, or you can contact the Department's national call centre who will record the pollution incident and route the complaint to the appropriate regional office of the Department.

2.1 LOCAL AUTHORITY / MUNICIPALITY

For pollution incidents that poses an imminent danger to human health, the environment or damage to property, contact the national toll free emergency number:

On a landline – 107
On a cell phone – 112

This will put you in touch with the closest emergency call centre and they should direct your call to the appropriate emergency response unit, the correct local authority department or government department.

If there is no imminent danger, contact the roads and stormwater department (if it involves pollution of the stormwater system and urban rivers) or the sewerage department (if it involves a leaking of surcharging sewage pipe) at your local municipality or city council. Their contact numbers would be in the local telephone directory.

2.1.1 Department of Water Affairs

To report pollution incidents that occur outside the boundaries of municipalities or city councils, contact the Department of Water Affairs national call centre toll free line or the nearest regional office of the Department of Water Affairs. The National Call Centre will register your call and will then refer it to the nearest regional DWA office to respond.

DEPARTMENT OF WATER AFFAIRS NATIONAL CALL CENTRE TOLL FREE LINE	
0800 200 200	
DEPARTMENT OF WATER AFFAIRS REGIONAL OFFICES	
EASTERN CAPE	
Private Bag X7485 KING WILLIAM'S TOWN 5600	Tel: (043) 604 5402 Fax: (043) 604 5592
FREE STATE	
PO Box 528 BLOEMFONTEIN 9300	Tel: (051) 405 9000 Fax: (051) 430 8146
GAUTENG	
Private Bag X995 PRETORIA 0001	Tel: (012) 392 1303 Fax: (012) 392 1304
KWAZULU-NATAL	
PO Box 1018 DURBAN 4000	Tel: (031) 336 2700 Fax: (031) 336 2849
MPUMALANGA	
Private Bag X11259 NELSPRUIT 1200	Tel: (013) 759 7310 Fax: (013) 759 7525

NORTH WEST	
Private Bag X5 MMABATHO 2735	Tel: (018) 387 9547 Fax: (018) 384 2059 Cell: 083 629 8991
NORTHERN CAPE	
Private Bag X6101 KIMBERLEY 8300	Tel: (053) 830 8804 Fax: (053) 830 8804
LIMPOPO PROVINCE	
Private Bag X9506 POLOKWANE 0700	Tel: (015) 295 1237 Fax: (015) 295 3217
WESTERN CAPE	
Private Bag X16 SANLAMHOF 7532	Tel: (021) 950 7100 Fax: (021) 946 366

2.2 WHAT INFORMATION TO SUPPLY

Please supply the following information when reporting a pollution incident:

Who	Supply your name, and contact details so that the authority that is responding to the incident to can contact you for more information if required.
What	Provide as much information as possible about the nature of the pollution incident. Describe what you saw such as: • the colour of the water and/or the discharge, • offensive odours (rotten egg, sour smell, sewage, chlorine, petrol fumes, dead fish), • was there foam present, • did the water have an oily sheen, • were there dead fish or dead aquatic organisms like frogs, • were there deposits on the stream or rivers banks, and • were there large amounts of algae or fungus present. If possible, take photographs of what you saw with your cell phone or camera.
Where	Describe where the pollution incident occurred and if possible, the physical extent of the area affected. If possible give the name of the stream, nearest town, street address, canals, nearby landmarks such as roads, bridges, easy to locate buildings, GPS coordinates, etc.
When	Describe when the incident was observed (date and time). Also indicate if the pollution was to your mind a single event or has the pollution occurred over an extended period of time.
How	Describe the possible cause of the pollution if it can be identified such as a leaking sewer, or tanker spill, etc.

2.2.1 Follow up

If possible follow up the incident report with a letter or email to the local authority or the Department of Water Affairs. It is also reasonable to expect the local authority or Department to give you feedback on their response to the pollution incident that you reported. If you

reported a pollution incident via the national DWA call centre, you will be given a reference number that can be used to track the response of the Department to the complaint.

PART 6 : REFERENCES

Department of Water Affairs and Forestry (DWAF), 2006. Resource Directed Management of Water Quality: Management Instruments. Volume 4.3: Guideline on Monitoring & Auditing for Resource Directed Management of Water Quality. Edition 2. Water Resource Planning Systems Series, Sub-Series No. WQP 1.7.3. ISBN N0. 0-621-36796-6. Department of Water Affairs and Forestry, Pretoria, South Africa.

DWAF: Resource Quality Services. 2004. Water Resource Quality Monitoring. Volume 1: Sampling Protocol for Inorganic Chemical Analysis.

DWAF: Resource Quality Services. 2004. Water Resource Quality Monitoring. Volume 2: Eutrophication Monitoring.

DWAF: Resource Quality Services. 2004. Water Resource Quality Monitoring. Volume 5: Fish Kill Investigation.

Gerber, A., Cilliers, C.J., van Ginkel, C. and Glen, R. 2004. Easy Identification of Aquatic Plants. ISBN: 0-621-3511-X. Department of Water Affairs & Forestry, Pretoria.

Michaud, J.P. 19991. A Citizen's Guide to Understanding and Monitoring Lands and Streams. http://www.ecy.wa.gov/programs/wq/plants/management/joysmanual/index.html

Myre, E. and Shaw, R. 2006. The Turbidity Tube: Simple and Accurate Measurement of Turbidity in the Field. www.cee.mtu.edu/sustainable_engineering/resources/technical/Turbidity-Myre_Shaw.pdf

Palmer, T., Berold, R., and Muller, N. (2004). *Environmental water quality in water resources management.* WRC Report TT 217/04. Water Research Commission, Pretoria.

Palmer, T., Berold, R., Muller, N., and Scherman, P. (2002). *Some, for all, forever. Water ecosystems and people.* WRC Report TT 176/02. Water Research Commission, Pretoria.

United States Department of Agriculture. Stream Visual Assessment Protocol. www.nrcs.usda.gov/technical/ECS/aquatic/svapfnl.pdf

United States Environmental Protection Agency. Volunteer Lake Monitoring: A Methods Manual. http://www.epa.gov/owow/monitoring/lakevm.html

United States Environmental Protection Agency. Volunteer Stream Monitoring: A Methods Manual. http://www.epa.gov/owow/monitoring/volunteer/stream/

Waterwatch Australia. Module 2: Getting Started: the team, monitoring and site. http://www.waterwatch.org.au/publications/module2/index.html

Waterwatch Australia. Module 3: Biological Parameters. http://www.waterwatch.org.au/publications/module3/index.html

Waterwatch Australia. Module 4: Physical and Chemical Parameters. http://www.waterwatch.org.au/publications/module4/index.html

APPENDIX 1

GENERIC WATER QUALITY LIMITS FOR VARIOUS WATER USER CATEGORIES

Generic water quality limits for various water user categories

In order to determine whether the water is suitable for its intended use, the fitness for use range has been developed for various water user categories. The following fitness for use categories are used:

Fitness for use category[6]	Definition
Ideal	The use of water is not affected in any way. The water is 100% fit for use by all users at all times.
Acceptable	Slight to moderate problems on a few occasions of for short periods of time.
Tolerable	Moderate to severe problems are encountered, usually for a limited period only.
Unacceptable	Water cannot be used for its intended use under normal circumstances at any time.

The generic water quality limits for various water user categories are provided in the Tables below. *NOTE: If the variable is not included in the Table, it means that this variable is not applicable to the water user. An expanded list of generic guidelines is available in DWAF (2006)[7].*

Table 1: Generic water quality limits for Domestic Use

Variable	Units	Fitness for use range			
		Ideal	Acceptable	Tolerable	Unacceptable
Turbidity	NTU	0 – 1	1 – 5	5 – 10	> 10
pH	pH units	5.0 – 9.5	5.0 – 4.5 9.5 – 10.0	4.5 – 4.0 10.0 – 10.5	< 4.0 > 10.5

Table 2: Generic water quality limits for Agricultural Use: Irrigation

Variable	Units	Fitness for use range			
		Ideal	Acceptable	Tolerable	Unacceptable
pH	pH units	6.5 – 8.4			< 6.5 > 8.4

Table 3: Generic water quality limits for Recreational Use

Variable	Units	Fitness for use range			
		Ideal	Acceptable	Tolerable	Unacceptable
Clarity	m	> 3	3.0 – 1.5	1.5 – 1.0	< 1.0
pH	pH units	6.5 – 8.5	6.5 – 5.0 8.5 – 9.0		0 – 5.0 > 9.0

[6] The fitness for use categories are colour coded for ease of reference.

[7] Department of Water Affairs and Forestry (DWAF), 2006. Resource Directed Management of Water Quality: Management Instruments. Volume 4.2. Guideline for Determining Resource Water Quality Objectives (RWQOs), Allocatable Water Quality and the Stress of the Water Resource. Resource Planning Systems Series, Sub-Series No. WQP 1.7.2. Department of Water Affairs and Forestry, Pretoria, South Africa.2006

Table 4: Generic water quality limits for Aquatic Ecosystems

Variable	Units	Fitness for use range			
		Ideal	**Acceptable**	**Tolerable**	**Unacceptable**
Temperature[8]	°C	Water temperature should not be allowed to vary from the ecosystems background average daily water temperature considered to be normal for that specific site and time of day, by > 2 °C, **or** by > 10 %, **whichever estimate is the more conservative**.			
DO	% of saturation	80 – 120 %	80 – 60 %	> 60 %	> 40 %
pH[9]		pH values should not be allowed to vary from the range of the background pH values for a specific site and time of day, by > 0.5 of a pH unit, **or** by > 5 %, **and should be assessed by whichever estimate is the more conservative**.			

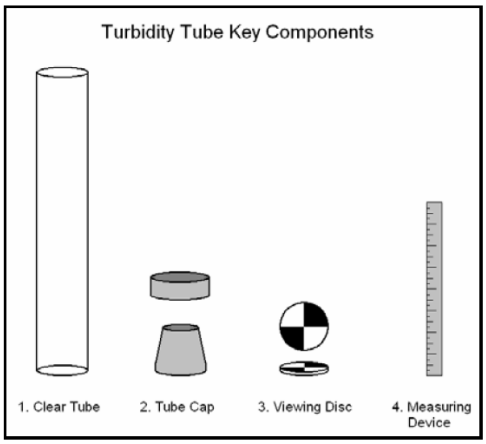
[8] The Ideal range for water temperature should be stated in terms of the case- and site-specific "natural" temperature regime. In all cases, local conditions should be determined before a water quality objective for an aquatic ecosystem is set. The 90th and 10th percentile values should be used to establish the "natural" temperature range.

[9] The Ideal range for pH should be stated in terms of the background site-specific pH regime. In all cases, local background conditions should be determined before a water quality objective for a particular aquatic ecosystem is set.

APPENDIX 2
HOW TO CONSTRUCT A TUBIDITY TUBE

Key components:

(1) ***A clear tube***. The clear tube will hold the water sample being tested. The tube must allow for maximum light reflectance off the marker being viewed.
(2) ***A tube cap***. The tube cap prevents the water sample from spilling out of the clear tube.
(3) ***A viewing disk***. The viewing disk will be submerged in the water sample. It is best to use a white background that is coloured with alternating black quadrants.
(4) ***Measuring device*** (i.e. ruler).



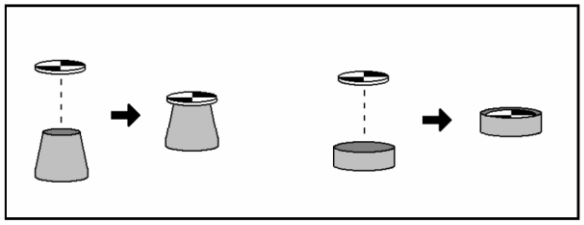
General construction:

Step 1: Plan the placement of the viewing disc

You will need to be able to see the viewing disk from the top of your clear tube. The placement of the viewing disc will depend on the tube cap. If the disc is not made from a floating material, it can be dropped to the bottom of the clear tube. Alternatively, you can attach the disc to the tube cap. Another possibility is to mark the tube cap with a checkered pattern so that it can be treated as a viewing disc.

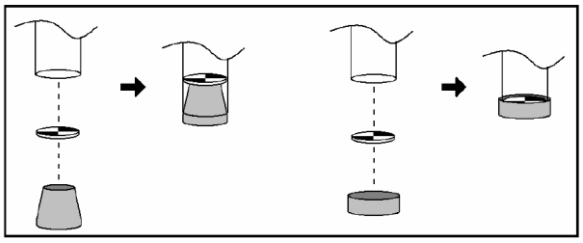
Step 2: Combine tube cap and viewing disc

You can use an adhesive or sealant to bind the viewing disc to the tube cap. Make sure that the disc will fit properly when the tube cap is inserted into the tube.



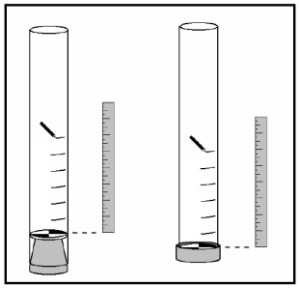
Step 3: Affix tube cap to bottom of the tube

Ideally the tube cap will be removable for cleaning, but the primary concern is that water does not escape the tube during testing. Make sure the disc is still clearly visible from the top of the tube.



Step 4: Mark tube with measurement increments

Ideally the turbidity level will be marked directly onto the tube. Place the zero mark of the measuring tape or ruler level with the viewing disc. Two rubber bands can be placed on each end of the measuring tape to hold it in place will you mark the levels with a marker.



Measurement procedure:

1. Dip the container/bucket into the water to collect the sample. Be careful not to include sediment from the bottom of the river.
2. Rinse the tube with the water that is going to be tested and pour it out.
3. Stir or swirl the water sample in the container vigorously until it is homogenous, introducing as little air as possible.
4. Place your head 10 – 20 cm directly over the tube so that you can see the viewing disk while the sample is being poured into the tube (steps 4 – 8 are illustrated in below).
5. Slowly pour water into the tube. Try not to form bubbles as your pour. If bubbles do form: stop pouring and allow any bubbles to rise and the surface of the water to become still.
6. Keep slowly adding water until the pattern on the disk becomes hard to see.
7. Stop pouring as soon as the pattern on the disk can no longer be seen.
8. Read the height of water in the turbidity tube and find the corresponding turbidity value in **Table 1**.
9. If you can still see the viewing disk pattern when the tube is full: record the turbidity value as greater than the final measuring mark (or less than 5 NTU).

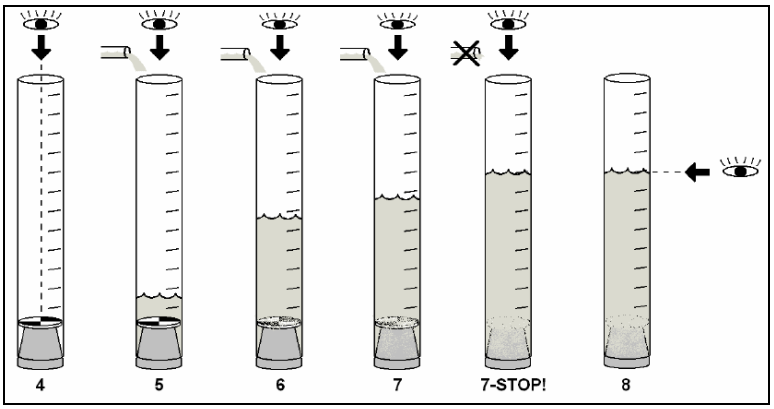


Table 1: Water-height to turbidity conversion

Centimetres	NTU
6.7	240
7.3	200
8.9	150
11.5	100
17.9	50
20.4	40
25.5	30
33.1	21
35.6	19
38.2	17
40.7	15
43.3	14
45.8	13
48.3	12
50.9	11
53.4	10
85.4	5

Myre, E. and Shaw, R. 2006. The Turbidity Tube: Simple and Accurate Measurement of Turbidity in the Field. www.cee.mtu.edu/sustainable_engineering/resources/technical/Turbidity-Myre_Shaw.pdf

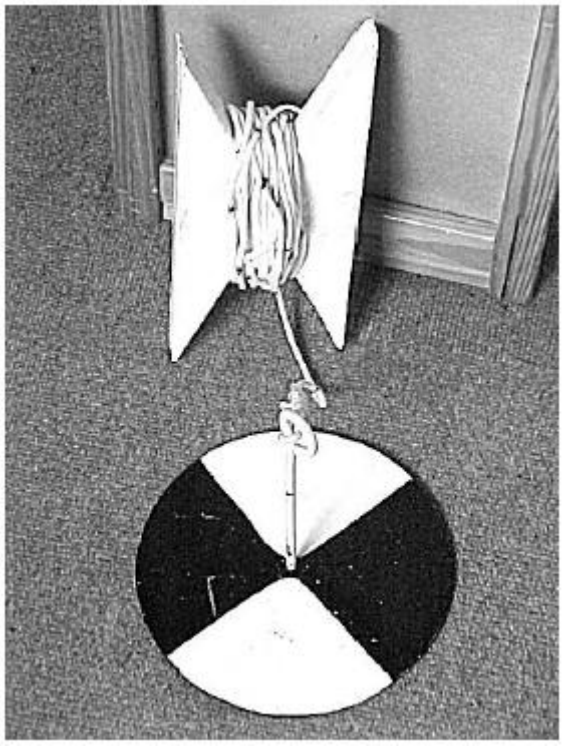
APPENDIX 3
HOW TO CONSTRUCT A SECCHI DISC

How to construct a Secchi disc:

A Secchi disc is used to determine the clarity of water in a reservoir. In order to assess the clarity, the Secchi disc is lowered into the water until the black and white quadrants are just visible. This depth of a measure of the clarity of the water.

Equipment:

The metal Secchi disc is 20 cm in diameter and is marked with alternative black and white quadrants. It is attached to a rope which is marked at 10 cm intervals. If the rope is not marked, a measuring tape can be used to determine the depth at which the Secchi disc is no longer visible.



Procedure:

(1) Lower the Secchi disc into the water, with the sun behind you, until it is out of site. NOTE: if the sun is in front of you, reflections off the surface of the water can influence the visibility of the disc.

(2) Lift the disc until it is just visible.

(3) Note and record the depth reading on the marked rope of using the measuring tape.